

ISSN 1713-7845

**JOURNAL**

*of the*  
**ENTOMOLOGICAL  
SOCIETY  
OF  
ONTARIO**



*Volume*  
*One Hundred and Forty Six*  
*2015*

Published 2015

**JOURNAL  
of the  
ENTOMOLOGICAL SOCIETY  
of  
ONTARIO**

*Volume One Hundred and Forty Six  
2015*

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**JOURNAL**  
*of the*  
**ENTOMOLOGICAL SOCIETY OF ONTARIO**

*VOLUME 146*

*2015*

This is my first volume as Editor of the Journal of the Entomological Society of Ontario. It is my great pleasure to take over the reins from John Huber. I would like to take this opportunity to thank John for his years of service to JESO, and for all his helpful advice over the past year as I have settled into the position.

It is quite a daunting task to take on the editorship of JESO at this period in its history. As most readers of this journal are aware, the fate of this publication has been up in the air over the past few years. Changes in the publication landscape in the recent past have resulted in an explosion of journals, and there has never been more choice available to authors as to where they can submit their work. This has had the rather unpleasant side effect of contributing to the reduction in the number of papers that are submitted to JESO. So where in that vast landscape of publication choices does this leave a small outpost like JESO? That is a question I hope to be able to answer during my tenure.

It is clear that if JESO is to survive it must adapt. We are fortunate that some of this work has begun and that we have a society and an executive that is supportive of further adaptations of JESO. We are also fortunate that there is a strong foundation of almost 150 years of publication upon which we can build. Under John's leadership, JESO has also begun some of the renovations necessary to bring us into the modern era, notably the adoption of an online-only publication and the archiving of the back catalogue with the Biodiversity Heritage Library. My task is to continue this work. My intent for 2016 is to complete the second stage of renovations to JESO by bringing us to an integrated submission and publication system that will allow our authors to submit papers using a modern web interface and allow our editorial team to more quickly review, edit, and publish your work. With the move to the online publication system, I also plan to begin the task of amalgamating JESO's back catalogue with the existing online offerings from the past few years. The long-term goal is that within the next five years the entire back catalogue of JESO will be online, searchable and indexed and available at one location.

My hope is that through this process we can raise the profile of JESO to the point that it becomes a first-choice for many authors. However, at the same time I am a realist. JESO will never challenge the large journals of the two major North American entomological societies. But that is okay. We can, though, become a journal of choice for regional contributions, particularly for works of taxonomy and systematics, and applied entomology, which have been traditional strengths for JESO. I also see JESO as a place where natural history about insects and arthropods can find a welcoming home. Moreover, I think JESO can serve as a home for those authors that have a passion for entomology,

even if entomology may not be their profession. These are potential authors who may never think of submitting to academic journals but still have important stories to tell. JESO may never win the war of the impact factors, but we can be a place where good work will find a welcome home.

So I end with what may be called the editor's plea: Send us your submissions! JESO has survived because of the support of its readers. I'm willing to bet that each of you has at least one paper that is looking for a welcome home but languishes at the back of your file cabinet (be it digital or physical). This is the kind of support that JESO needs as we begin to adapt to the new reality. During this period we also ask that you liken us to your favourite local establishment: We may be under new management and the sign out front says 'Under Renovations' but we still want your business and are no less dedicated to your satisfaction. Please stick with us while we go through this process. It will be worth it. Whereas in previous years publications were made annually and undesirably after the NSERC application cycle, articles can now be posted online instantly upon acceptance. This is a major benefit to graduate students in particular who wish to submit their work for publication prior to applying for scholarships. The other major benefit is that we are now an online open access publication, and do not penalize authors for submitting large numbers of pages or images. Consider us as you contemplate on where to submit those lengthy taxonomic revisions.

Before I go, I would like to end with a thank you to the associate editors of JESO and the reviewers who gave their time to help us with this issue. I would also like to thank Neil Carter for his service to JESO over the years and welcome Jocelyn Smith as a new associate editor. Technical editing of JESO is done by Tom Onuferko. Together these folks have helped put together the following 54 pages. I hope you enjoy reading them. Submissions for volume 147 are now being accepted and can be sent to [JESOEditor@gmail.com](mailto:JESOEditor@gmail.com).

Chris MacQuarrie  
Editor

**A LIST OF BEE SPECIES (HYMENOPTERA: APOIDEA)  
RECORDED FROM THREE MUNICIPALITIES IN THE NIAGARA  
REGION OF ONTARIO, INCLUDING A NEW RECORD OF  
*LASIOGLOSSUM FURUNCULUM* GIBBS (HALICTIDAE) IN  
CANADA**

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**Abstract**

*J. ent. Soc. Ont.* 146: 3–22

We carried out an extensive survey of bee species in the Niagara Region of Ontario, Canada, by sampling various sites within three municipalities from 2003 to 2013. The municipalities were St. Catharines, Port Colborne, and Wainfleet. Sampling mainly consisted of pan-trapping, but also included sweeping through vegetation and targeted collection from flowers. In the longest ongoing survey of a bee community to date in Canada, we collected 51,842 bee specimens comprising nearly 150 valid species, of which 30 were not previously recorded for the region. We also present the first record of the rare sweat bee species *Lasioglossum furunculum* Gibbs (Hymenoptera: Halictidae) in Canada, which was previously known only from Massachusetts, United States of America.

*Published November 2015*

**Introduction**

Our first survey of a bee community in the Niagara Region of southern Ontario, Canada, was carried out in 2003 at 8 sites on the Brock University campus and the adjacent Glenridge Quarry Naturalization Site in St. Catharines in the northeastern tier of the Niagara Peninsula (43.1 °N, 79.2 °W; Richards *et al.* 2011). The St. Catharines sites included relatively undisturbed meadows and fields on the Brock University campus, as well as regeneration sites at the Glenridge Quarry Naturalization Site, a former landfill.

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Descriptions of the St. Catharines sites and the history of the area (natural and in terms of human activity) were provided in Richards *et al.* (2011). The St. Catharines sites were sampled each year from 2004 until 2013, except 2007, for a total of ten years of sampling. From 2011 to 2013, bees were also systematically sampled at two landfill regeneration sites in southern Niagara Peninsula, the Elm Street Naturalization Site in Port Colborne, Ontario (42.9 °N, 79.3 °W), and the Station Road Naturalization Site in Wainfleet, Ontario (42.9 °N, 79.4 °W). The sites in Port Colborne and Wainfleet are located on sites that from the 1950s until 2009 and 2008, respectively, functioned as municipal landfills. By 2011, these landfills had been capped with clay, covered with soil, and planted with an array of flowering plant species native to North America.

Our objective in the present study is to provide the list of bee species collected from our sites in these three municipalities of the Niagara Region. All three sampling areas are within the Carolinian Zone, which includes tallgrass prairie and woodland communities. Considering the proximity (< 30 km) of the sites and the longer sampling effort at St. Catharines, we expected that the species lists compiled for the Port Colborne and Wainfleet municipalities would be subsets of the St. Catharines list.

## Methods

Bees were collected using three methods: pan-traps (2003–2006, 2008–2013), sweep-netting vegetation (2003–2005), and aerial netting from flowers (2003–2005, 2011–2013). Details on sampling methodology are provided in Richards *et al.* (2011) and Rutgers-Kelly and Richards (2013). While combining specimens from all these collecting methods maximized the number of species likely to be collected (Wilson *et al.* 2008), non-standard sampling across years and sites means that it is difficult to quantify and compare the proportional representations of bee species in the community.

All specimens were pinned and labelled, and are currently deposited in the research collection of M. H. Richards at Brock University. The majority of specimens collected in St. Catharines after 2003 were identified by T. M. Onuferko, and those collected in Port Colborne and Wainfleet from 2011–2013 were identified by R. Kutby and T. M. Onuferko. The following taxonomic keys were used to identify specimens in conjunction with online keys available on Discover Life (Ascher and Pickering 2015): Colla *et al.* (2011) for *Bombus* Latreille (Hymenoptera: Apidae); Gibbs (2010, 2011) for *Lasioglossum* Curtis (Hymenoptera: Halictidae) subgenus *Dialictus* Robertson; Gibbs *et al.* (2013) for *Lasioglossum* subgenera *Evyllaesus* Robertson, *Hemihalictus* Cockerell, and *Sphecodogastra* Ashmead; Mitchell (1960, 1962) for *Halictus* Latreille (Hymenoptera: Halictidae) and *Sphecodes* Latreille (Hymenoptera: Halictidae); McGinley (1986) for *Lasioglossum* subgenera *Lasioglossum* and *Leuchalictus* Warncke; Rehan and Richards (2008) and Rehan and Sheffield (2011) for *Ceratina* Latreille (Hymenoptera: Apidae); Rightmyer (2008) for *Triepeolus* Robertson (Hymenoptera: Apidae); and Sheffield *et al.* (2011b) for *Megachile* Latreille (Hymenoptera: Megachilidae). Discover Life keys (Ascher and Pickering 2015) were used for the following genera: *Agapostemon* Guérin-Ménéville (Hymenoptera: Apidae), *Anthidium* Fabricius (Hymenoptera: Megachilidae), *Anthophora* Latreille (Hymenoptera: Apidae), *Calliopsis* Smith (Hymenoptera: Andrenidae), *Chelostoma* Latreille (Hymenoptera: Megachilidae),

*Coelioxys* Latreille (Hymenoptera: Megachilidae), *Heriades* Spinola (Hymenoptera: Megachilidae), *Hoplitis* Klug (Hymenoptera: Megachilidae), *Hylaeus* Fabricius (Hymenoptera: Colletidae), *Melissodes* Latreille (Hymenoptera: Apidae), *Osmia* Panzer (Hymenoptera: Megachilidae), and *Stelis* Panzer (Hymenoptera: Megachilidae). Females of the following species pairs are very difficult to differentiate, and identifications were largely based on male characters: *Ceratina dupla* Say versus *C. mikmaqi* Rehan and Sheffield, and *Hylaeus affinis* (Smith) versus *H. modestus* Say. Specimens of *Nomada* Scopoli (Hymenoptera: Apidae), a genus in need of revision, were kindly identified by Sam Droege (US Geological Survey, Patuxent Wildlife Research Center, Beltsville, Maryland). All *Andrena* Fabricius (Hymenoptera: Andrenidae) designations were made by Cory Sheffield (Royal Saskatchewan Museum, Regina, Saskatchewan), for which we are most grateful, and Jason Gibbs (Michigan State University, East Lansing, Michigan) graciously helped to identify many of the *Lasioglossum* specimens, including one new record for Canada.

## Results

A total of 51,842 bee specimens were collected, comprising 149 species and 1 morphospecies of *Nomada*. Richards *et al.* (2011) had previously identified 124 species and morphospecies from the 2003 samples, including four distinct morphospecies and one unknown species of *Nomada*. In the present study, these *Nomada* have been collapsed into a single bidentate morphospecies group, as suggested by taxonomic expert Sam Droege. In the present study, 30 valid species not recorded by Richards *et al.* (2011) were identified (see Table 1 for a list of these and all other species sampled). All species belonged to the five most common bee families occurring in North America (Andrenidae, Apidae, Colletidae, Halictidae, and Megachilidae); no bees of the small and uncommon family Melittidae were sampled or observed. Of the 30 bee genera represented, only the cleptoparasitic genus *Triepeolus* was not previously recorded by Richards *et al.* (2011). Almost a third (9/30) of the new species added are cleptoparasitic or socially parasitic, one of which is described for the first time in Canada in the section that follows. The most speciose family sampled was Halictidae (54 species), and the least speciose was Colletidae (11 species). The present ranking of families by morphospecies richness (Halictidae > Apidae > Megachilidae > Andrenidae > Colletidae) generally matches that of Richards *et al.* (2011) (Halictidae > Apidae = Megachilidae > Andrenidae > Colletidae).

### **New record for Canada: *Lasioglossum (Dialictus) furunculum* Gibbs**

*Lasioglossum furunculum* is a species that was recently described from Massachusetts, United States of America, from a single specimen (Gibbs 2011). It is most similar to *Lasioglossum izawsum* Gibbs (Hymenoptera: Halictidae), but differs in having no preapical tooth on the mandible (Fig. 1A) and an inner metatibial spur with four rather than three branches (Gibbs 2011). Females of another similar species, *Lasioglossum simplex* (Robertson) (Hymenoptera: Halictidae), lack a carinate pronotal ridge present in the two abovementioned species (Gibbs 2011). In all three species, the gena is subequal in width to the compound eye when viewed from the side (Fig. 1B); it is conspicuously wider in other parasitic species (Gibbs 2011).

TABLE 1: A complete checklist of bee species sampled from 2003–2013 in southern St. Catharines, in Port Colborne, and in Wainfleet, Ontario, Canada. Species collected since the initial 2003 survey (Richards *et al.* 2011) are indicated by an asterisk (\*). Species for which a life history trait is suspected but not confirmed, as in *Lasiglossum* spp., are indicated by a question mark (?). The presence of a species within a particular municipality is denoted with an ‘X’. Foraging habit is listed as N/A for parasitic species, which do not forage.

Family and species	Life History trait			Origin	Municipality		
	Social habit	Nesting habit	Foraging habit		St. Catharines	Port Colborne	Wainfleet
Andrenidae (20 species)							
Andrena (Andrena) mandibularis	Robertson	Solitary	Ground	Generalist	Native	X	
Andrena (Andrena) thaspiae	Graenicher	Solitary	Ground	Generalist	Native	X	
Andrena (Euandrena) nigrihirta	(Ashmead)	Solitary	Ground	Generalist	Native	X	
*Andrena (Gonandrena) fragilis	Smith	Solitary	Ground	Cornus Linnaeus (Cornaceae)	Native	X	
*Andrena (Gonandrena) integra	Smith	Solitary	Ground	Cornus Linnaeus (Cornaceae)	Native	X	
Andrena (Holandrena) cressonii	Robertson	Solitary	Ground	Generalist	Native	X	
Andrena (Larandrena) miserabilis	Cresson	Solitary	Ground	Generalist	Native	X	
Andrena (Leucandrena) erythronii	Robertson	Solitary	Ground	Erythronium Linnaeus (Liliaceae)	Native	X	
Andrena (Melandrena) carlini	Cockerell	Solitary	Ground	Generalist	Native	X	
Andrena (Melandrena) dunmingi	Cockerell	Solitary	Ground	Generalist	Native	X	
*Andrena (Melandrena) vicina	Smith	Solitary	Ground	Generalist	Native	X	X
Andrena (Ptilandrena) erigeniae	Robertson	Solitary	Ground	Claytonia Linnaeus (Montiaceae)	Native	X	X
Andrena (Scaphandrena) arabis	Robertson	Solitary	Ground	Brassicaceae	Native	X	
Andrena (Simandrena) nasonii	Robertson	Solitary	Ground	Generalist	Native	X	
Andrena (Simandrena) wheeleri	Graenicher	Solitary	Ground	Generalist	Native	X	
Andrena (Simandrena) wilkella	(Kirby)	Solitary	Ground	Fabaceae?	Native	X	X



TABLE 1 continued...

Family and species	Life History trait		Origin	Municipality		
	Social habit	Nesting habit		Foraging habit	St. Catharines	Port Colborne
<i>Andrena (Trachandrena) forbesii</i> Robertson	Solitary	Ground	Native	Generalist	X	
<i>Andrena (Trachandrena) hippotes</i> Robertson	Solitary	Ground	Native	Generalist	X	
<i>Calliopsis (Calliopsis) andreniformis</i> Smith	Solitary	Ground	Native	Generalist	X	X
<i>Pseudopanurgus andreoides</i> (Smith)	Solitary	Ground	Native	Asteraceae	X	
<b>Apidae (33 species and morphospecies)</b>						
<i>Anthophora (Clisodon) terminalis</i> Cresson	Solitary	Stems/wood	Native	Generalist	X	X
* <i>Anthophora (Melea) bomboidea</i> Kirby	Solitary	Ground	Native	Generalist	X	X
<i>Apis (Apis) mellifera</i> Linnaeus	Eusocial	Cavities	Exotic	Generalist	X	X
* <i>Bombus (Bombus) terricola</i> Kirby	Eusocial	Underground	Native	Generalist	X	X
<i>Bombus (Cullumanobombus) griseocollis</i> (DeGeer)	Eusocial	Ground surface	Native	Generalist	X	X
<i>Bombus (Cullumanobombus) rufocinctus</i> Cresson	Eusocial	Ground surface or underground	Native	Generalist	X	X
<i>Bombus (Psithyrus) citrinus</i> (Smith)	Social parasite	Nests of <i>B. impatiens</i> and <i>B. vagans</i>	Native	N/A	X	
<i>Bombus (Pyrobombus) bimaculatus</i> Cresson	Eusocial	Underground	Native	Generalist	X	X
<i>Bombus (Pyrobombus) impatiens</i> Cresson	Eusocial	Underground	Native	Generalist	X	X
<i>Bombus (Pyrobombus) ternarius</i> Say	Eusocial	Underground	Native	Generalist	X	
<i>Bombus (Pyrobombus) vagans</i> Smith	Eusocial	Ground surface or underground	Native	Generalist	X	X
* <i>Bombus (Subterraneobombus) borealis</i> Kirby	Eusocial	Underground	Native	Generalist	X	X

TABLE 1 continued...

Family and species	Life History trait			Origin	Municipality		
	Social habit	Nesting habit	Foraging habit		St. Catharines	Port Colborne	Wainfleet
<i>Bombus (Thoracobombus) fervidus</i> (Fabricius)	Eusocial	Ground surface	Generalist	Native	X	X	X
<i>Ceratina (Zadontomerus) calcarata</i> Robertson	Solitary	Stems	Generalist	Native	X	X	X
<i>Ceratina (Zadontomerus) dupla</i> Say	Solitary	Stems	Generalist	Native	X	X	X
* <i>Ceratina (Zadontomerus) mikmaqi</i> Rehan and Sheffield	Solitary	Stems	Generalist	Native	X	X	X
<i>Ceratina (Zadontomerus) strenua</i> Smith	Solitary	Stems	Generalist	Native	X		
<i>Melissodes (Apomelissodes) apicata</i> Lovell and Cockerell	Solitary	Ground	<i>Pontederia cordata</i> Linnaeus (Pontederiaceae) Asteraceae	Native	X		
<i>Melissodes (Eumelissodes) druriella</i> (Kirby)	Solitary	Ground		Native	X		X
<i>Melissodes (Helimelissodes) desponsa</i> Smith	Solitary	Ground	<i>Cirsium</i> Miller (Asteraceae)	Native	X	X	X
<i>Nomada articulata</i> Smith	Cleptoparasite	Nests of <i>Agapostemon</i> Unknown	N/A	Native	X		
<i>Nomada bethunei</i> Cockerell	Cleptoparasite		N/A	Native	X		
* <i>Nomada cressonii</i> Robertson	Cleptoparasite	Nests of <i>Andrena</i> , including <i>A. crataegi</i> (not collected)	N/A	Native	X		
<i>Nomada denticulata</i> Robertson	Cleptoparasite	Nests of <i>Andrena</i>	N/A	Native	X		
<i>Nomada illinoensis/sayi</i> Robertson	Cleptoparasite	Nests of <i>Andrena</i>	N/A	Native	X		
* <i>Nomada imbricata</i> Smith	Cleptoparasite	Nests of <i>Andrena</i>	N/A	Native	X		
* <i>Nomada luteoloides</i> Robertson	Cleptoparasite	Nests of <i>Andrena</i>	N/A	Native	X		

TABLE 1 continued...

Family and species	Life History trait		Origin	Municipality		
	Social habit	Nesting habit		Foraging habit	St. Catharines	Port Colborne
* <i>Nomada maculata</i> Cresson	Cleptoparasite	Nests of <i>Andrena vicina</i>	Native	N/A	X	
		Unknown				
<i>Nomada pygmaea</i> Cresson	Cleptoparasite	Unknown	Native	N/A	X	
<i>Nomada bidentata</i> group	Cleptoparasite	Unknown	Native	N/A	X	
* <i>Triepeolus donatus</i> (Smith)	Cleptoparasite	Nests of <i>Melissodes desponsa</i>	Native	N/A	X	
* <i>Triepeolus lunatus</i> (Say)	Cleptoparasite	Nests of <i>Melissodes</i>	Native	N/A	X	
<i>Xylocopa (Xylocopoides) virginica</i> (Linnaeus)	Social	Wood	Native	Generalist	X	X
<b>Colletidae (11 species)</b>						
<i>Colletes americanus</i> Cresson	Solitary	Ground	Native	Asteraceae	X	
<i>Colletes compactus</i> Cresson	Solitary	Ground	Native	Asteraceae	X	
<i>Colletes simulans</i> Cresson	Solitary	Ground	Native	Asteraceae	X	
<i>Hylaeus (Hylaeus) annulatus</i> (Linnaeus)	Solitary	Stems/cavities	Native	Generalist	X	X
<i>Hylaeus (Hylaeus) mesillae</i> (Cockerell)	Solitary	Stems/cavities	Native	Generalist	X	X
* <i>Hylaeus (Metziella) sparsus</i> (Cresson)	Solitary	Cavities	Native	Apiaceae	X	
<i>Hylaeus (Prosopis) affinis</i> (Smith)	Solitary	Stems/cavities	Native	Generalist	X	X
<i>Hylaeus (Prosopis) illinoisensis</i> (Robertson)	Solitary	Stems	Native	Generalist	X	
<i>Hylaeus (Prosopis) modestus</i> Say	Solitary	Stems/cavities	Native	Generalist	X	X
<i>Hylaeus (Spatulariella) hyalinatus</i> Smith	Solitary	Stems	Exotic	Generalist	X	X
* <i>Hylaeus (Spatulariella) punctatus</i> (Brullé)	Solitary	Pre-existing cavities	Exotic	Generalist?	X	

TABLE 1 continued ...

Family and species	Life History trait			Origin	Municipality		
	Social habit	Nesting habit	Foraging habit		St. Catharines	Port Colborne	Wainfleet
Halictidae (54 species)							
*Agapostemon (Agapostemon) texanus Cresson	Solitary	Ground	Generalist	Native	X		
Agapostemon (Agapostemon) virescens (Fabricius)	Communal	Ground	Generalist	Native	X	X	X
Augochlora (Augochlora) pura (Say)	Solitary	Wood	Generalist	Native	X	X	X
Augochlarella aurata (Smith)	Eusocial	Ground	Generalist	Native	X	X	X
Augochloropsis (Paraugochloropsis) metallica (Fabricius)	Semisocial?	Ground	Generalist	Native	X		
Dufourea monardae (Viereck)	Solitary	Ground	Monarda Linnaeus	Native	X		
Halictus (Odontalictus) ligatus Say	Eusocial	Ground	(Lamiaceae) Generalist	Native	X	X	X
Halictus (Protohalictus) rubicundus (Christ)	Solitary/eusocial	Ground	Generalist	Native	X	X	X
Halictus (Seladonia) confusus Smith	Solitary/eusocial	Ground	Generalist	Native	X	X	X
Lasioglossum (Dialictus ) admirandum (Sandhouse)	Eusocial?	Ground	Generalist	Native	X	X	X
Lasioglossum (Dialictus) albipenne (Robertson)	Eusocial?	Ground	Generalist	Native	X		
Lasioglossum (Dialictus) atwoodi Gibbs	Eusocial?	Ground	Generalist	Native	X	X	X
Lasioglossum (Dialictus) coeruleum (Robertson)	Eusocial	Wood	Generalist	Native	X		
Lasioglossum (Dialictus) cressonii (Robertson)	Eusocial?	Wood	Generalist	Native	X	X	X
Lasioglossum (Dialictus) dreisbachi (Mitchell)	Unknown	Unknown	Salix Linnaeus	Native	X		
Lasioglossum (Dialictus) ellisiae (Sandhouse)	Eusocial?	Ground	(Salicaceae) Generalist	Native	X		
Lasioglossum (Dialictus) ephialtum Gibbs	Eusocial?	Ground	Generalist	Native	X	X	X



TABLE 1 continued...

Family and species	Life History trait		Origin	Municipality		
	Social habit	Nesting habit		Foraging habit	St. Catharines	Port Colborne
<i>Lasioglossum (Dialictus) fattigi</i> (Mitchell)	Eusocial?	Ground	Native	Generalist	X	X
* <i>Lasioglossum (Dialictus) furunculum</i> Gibbs	Clepto- or social parasite	Nests of nest-building <i>Dialictus</i> ?	Native	N/A	X	
<i>Lasioglossum (Dialictus) hitchensi</i> Gibbs	Eusocial?	Ground	Native	Generalist	X	X
<i>Lasioglossum (Dialictus) imitatum</i> (Smith)	Eusocial	Ground	Native	Generalist	X	X
<i>Lasioglossum (Dialictus) laevissimum</i> (Smith)	Eusocial	Ground	Native	Generalist	X	X
<i>Lasioglossum (Dialictus) leucocomum</i> (Lovell)	Eusocial?	Ground	Native	Generalist	X	X
<i>Lasioglossum (Dialictus) lineatulum</i> (Crawford)	Eusocial	Ground	Native	Generalist	X	X
<i>Lasioglossum (Dialictus) lionotum</i> (Sandhouse)	Social parasite	Nests of <i>L. imitatum</i>	Native	N/A	X	
<i>Lasioglossum (Dialictus) michiganense</i> (Mitchell)	Clepto- or social parasite	Nests of nest-building <i>Dialictus</i> ?	Native	N/A	X	
<i>Lasioglossum (Dialictus) nigroviride</i> (Graenicher)	Eusocial?	Wood	Native	Generalist	X	X
* <i>Lasioglossum (Dialictus) oblongum</i> (Lovell)	Unknown	Wood	Native	Generalist	X	X
<i>Lasioglossum (Dialictus) oceanicum</i> (Cockerell)	Eusocial?	Ground	Native	Generalist	X	
<i>Lasioglossum (Dialictus) paradmirationum</i> (Knerer and Atwood)	Eusocial?	Ground	Native	Generalist	X	X
<i>Lasioglossum (Dialictus) perpunctatum</i> (Ellis)	Eusocial?	Ground	Native	Generalist	X	
* <i>Lasioglossum (Dialictus) pilosum</i> (Smith)	Communal?	Ground	Native	Generalist		X
<i>Lasioglossum (Dialictus) planatum</i> (Lovell)	Eusocial?	Ground?	Native	Generalist	X	
* <i>Lasioglossum (Dialictus) platyparium</i> (Robertson)	Social parasite	Nests of nest-building <i>Dialictus</i> ?	Native	N/A	X	

TABLE 1 continued...

Family and species	Life History trait			Origin	Municipality		
	Social habit	Nesting habit	Foraging habit		St. Catharines	Port Colborne	Wainfleet
<i>Lasioglossum (Dialictus) sagax</i> (Sandhouse)	Eusocial?	Ground	Generalist	Native	X	X	
<i>Lasioglossum (Dialictus) smilacinae</i> (Robertson)	Eusocial?	Ground	Generalist	Native	X		
* <i>Lasioglossum (Dialictus) subviridatum</i> (Cockerell)	Unknown	Wood	Generalist	Native	X		
* <i>Lasioglossum (Dialictus) versans</i> (Lovell)	Unknown	Ground	Generalist	Native	X		
<i>Lasioglossum (Dialictus) versatum</i> (Robertson)	Eusocial	Ground	Generalist	Native	X	X	X
<i>Lasioglossum (Dialictus) viridatum</i> (Lovell)	Eusocial	Ground	Generalist	Native	X	X	X
* <i>Lasioglossum (Dialictus) weemsi</i> (Mitchell)	Eusocial?	Ground?	Generalist?	Native	X	X	X
<i>Lasioglossum (Dialictus) zephyrum</i> (Smith)	Eusocial	Ground	Generalist	Native	X	X	X
<i>Lasioglossum (Evyllaesus) cinctipes</i> (Provancher)	Eusocial	Ground	Generalist	Native	X	X	
<i>Lasioglossum (Hemihalictus) foxii</i> (Robertson)	Solitary?	Ground	Generalist	Native	X		
<i>Lasioglossum (Hemihalictus) inconditum</i> (Cockerell)	Solitary?	Ground	Generalist	Native	X		
<i>Lasioglossum (Hemihalictus)</i> <i>macoupinense</i> (Robertson)	Solitary	Ground	Generalist	Native	X		
<i>Lasioglossum (Lasioglossum) coriaceum</i> (Smith)	Solitary	Ground	Generalist	Native	X	X	
<i>Lasioglossum (Leuchalictus) leucozonium</i> (Schrack)	Solitary	Ground	Generalist	Exotic	X	X	X
<i>Lasioglossum (Leuchalictus) zomilum</i> (Smith)	Solitary	Ground	Generalist	Exotic	X	X	X
* <i>Lasioglossum (Sphecodogastra)</i> <i>quebecense</i> (Crawford)	Solitary	Ground	Generalist	Native	X		
* <i>Sphecodes atlantis</i> Mitchell	Cleptoparasite	Nests of nest-building <i>Lasioglossum</i>	N/A	Native	X	X	

TABLE 1 continued...

Family and species	Life History trait		Origin	Municipality		
	Social habit	Nesting habit		Foraging habit	St. Catharines	Port Colborne
<i>Sphecodes dichrous</i> Smith	Cleptoparasite	Nests of nest-building <i>Halictus</i> and/or <i>Lasiosomus</i>	Native	N/A	X	
<i>Sphecodes heraclei</i> Robertson	Cleptoparasite	Nests of nest-building <i>Halictus</i> and/or <i>Lasiosomus</i>	Native	N/A	X	
<i>Sphecodes ranunculi</i> Robertson	Cleptoparasite	Nests of nest-building <i>Halictus</i> and/or <i>Lasiosomus</i>	Native	N/A	X	
<b>Megachilidae (32 species)</b>						
<i>Anthidiellum (Loyolanthidium) notatum</i> (Latreille)	Solitary	Makes nests on surfaces	Native	Generalist	X	
<i>Anthidium (Anthidium) manicatum</i> (Linnaeus)	Solitary	Cavities	Exotic	Generalist	X	X
* <i>Anthidium (Proanthidium) oblongatum</i> (Illiger)	Solitary	Cavities	Exotic	Generalist	X	
<i>Chelostoma (Gyrodromella) ranunculi</i> (Lepeletier)	Solitary	Cavities	Exotic	<i>Campanula</i> Linnaeus (Campanulaceae)	X	
<i>Coelioxys (Boreocoelioxys) octodentata</i> Say	Cleptoparasite	Nests of <i>Megachile brevis</i> , <i>M. centuncularis</i> , and <i>M. mendica</i>	Native	N/A	X	X

TABLE 1 continued...

Family and species	Life History trait			Origin	Municipality		
	Social habit	Nesting habit	Foraging habit		St. Catharines	Port Colborne	Wainfleet
<i>Coelioxys (Boreocoelioxys) rufitarsis</i> Smith	Cleptoparasite	Nests of <i>Megachile latimanus</i> and <i>M. melanophaea</i> Smith (not collected)	N/A	Native	X		
<i>Heriades (Neotrypetes) leavittii</i> Crawford	Solitary	Cavities	Generalist	Native	X		
<i>Heriades (Neotrypetes) variolosa</i> (Cresson)	Solitary	Cavities	Generalist	Native	X		
<i>Heriades (Neotrypetes) carinata</i> Cresson	Solitary	Cavities	Generalist	Native	X	X	
<i>Hoplitis (Alcidamea) pilosifrons</i> (Cresson)	Solitary	Cavities	Generalist	Native	X	X	X
<i>Hoplitis (Alcidamea) producta</i> (Cresson)	Solitary	Cavities	Generalist	Native	X	X	
<i>Hoplitis (Alcidamea) spoliata</i> (Provancher)	Solitary	Cavities	Generalist	Native	X	X	
* <i>Megachile (Callomegachile) sculpturalis</i> Smith	Solitary	Cavities	Generalist	Exotic	X		X
<i>Megachile (Chelostomoides) campanulae</i> (Robertson)	Solitary	Cavities	Generalist	Native	X		
<i>Megachile (Eutricharaea) rotundata</i> (Fabricius)	Solitary	Cavities	Generalist	Exotic	X	X	X
<i>Megachile (Litomegachile) brevis</i> Say	Solitary	Cavities/ground	Generalist	Native	X	X	X
<i>Megachile (Litomegachile) mendica</i> Cresson	Solitary	Cavities/ground	Generalist	Native	X	X	
<i>Megachile (Litomegachile) texana</i> Cresson	Solitary	Ground	Generalist	Native	X		
<i>Megachile (Megachile) centuncularis</i> (Linnaeus)	Solitary	Cavities	Generalist	Possibly exotic	X	X	X
<i>Megachile (Megachile) relativa</i> Cresson	Solitary	Cavities	Generalist	Native	X		X
<i>Megachile (Pseudomegachile) ericetorum</i> Lepeletier	Solitary	Cavities	Generalist	Exotic	X	X	



TABLE 1 continued...

Family and species	Life History trait		Origin	Municipality		
	Social habit	Nesting habit		Foraging habit	St. Catharines	Port Colborne
<i>Megachile (Sayapis) pugnata</i> Say	Solitary	Cavities	Native	Generalist	X	
* <i>Megachile (Xanthosarus) frigida</i> Smith	Solitary	Wood	Native	Generalist	X	
* <i>Megachile (Xanthosarus) gemula</i> Cresson	Solitary	Ground	Native	Generalist	X	
<i>Megachile (Xanthosarus) latimanus</i> Say	Solitary	Ground	Native	Generalist	X	X
<i>Osmia (Diceratosmia) conjuncta</i> Cresson	Solitary	Snail shells	Native	Generalist	X	X
* <i>Osmia (Helicosmia) georgica</i> Cresson	Solitary	Wood	Native	Asteraceae specialist		X
<i>Osmia (Melanosmia) atriventris</i> Cresson	Solitary	Cavities	Native	Generalist	X	X
<i>Osmia (Melanosmia) pumila</i> Cresson	Solitary	Stems/cavities	Native	Generalist	X	X
<i>Osmia (Melanosmia) simillima</i> Smith	Solitary	Wood	Native	Generalist	X	
<i>Osmia (Osmia) lignaria lignaria</i> Say	Solitary	Cavities	Native	Generalist	X	
<i>Stelis (Stelis) lateralis</i> Cresson	Cleptoparasite	Nests of <i>Hoplitis pilosifrons</i> , <i>H. producta</i> , <i>H. simplex</i> (not collected)	Native	N/A	X	X

Notes:

- 1) The following species were listed under different names in Richards *et al.* (2011) (their taxonomy has since changed), with previous designations provided in parentheses: *Pseudopanurgus andrenoides* (*Protandrena andrenoides*), *Ceratina mikmaqi* (*Ceratina dupla* [in part]), *Nomada bidentate* group (Forms D, H, N, and O [in part]), *Lasioglossum hitchensi* (*Lasioglossum mitchelli* Gibbs), *Lasioglossum lionotum* (*Lasioglossum asteris* (Mitchell)), *Lasioglossum smilacinae* (*Lasioglossum zophops* (Robertson)), *Lasioglossum inconditum* (*Lasioglossum rufitarse* (Zetterstedt)), *Lasioglossum macoupinense* (*Lasioglossum divergens* (Lovell)), *Heriades variolosa* (*Heriades variolosus*), *Heriades carinata* (*Heriades carinatus*).
- 2) Bidentate species of *Nomada* are in need of revision, and those in our collection may include several undescribed species, but here we treat bidentate forms as a single morphospecies.
- 3) Life history information was taken from the following sources: Arduser (2002), Ascher (2001), Ascher *et al.* (2014), Bohart and Nye (1956), Bouseman and LaBerge (1978), Colla *et al.* (2011), Eickwort and Abrams (1980), Gibbs (2010, 2011), Gibbs *et al.* (2013), Grixti and Packer (2006), Hartman *et al.* (1944), LaBerge (1973, 1980, 1985, 1987, 1989), LaBerge and Ribble (1972, 1975), Michener (2007), Mitchell (1960, 1962), Osgood (1989), Packer *et al.* (2007), Ribble (1967, 1974), Richards *et al.* (2011), Rightmyer (2008), Roberts (1973), Sheffield *et al.* (2010, 2011a, 2011b, 2014), Stockhammer (1967), Taraday (1982), and Wolf and Ascher (2009).

We collected a single female specimen of *L. furunculum* on 9 September 2009 from St. Catharines on the periphery of the Glenridge Quarry Naturalization Site, just south of the Niagara Escarpment. Males of the species are unknown (Gibbs 2011). Given the similarity of this species to *L. izawsum*, the lack of DNA barcodes, and the limited number of individuals available for both species, it is possible that *L. furunculum* and *L. izawsum* are synonymous (Gibbs 2011). However, the two species are currently recognized as valid, and the St. Catharines specimen best matches the description of *L. furunculum*. The species is presumably a cleptoparasite or a social parasite of one or more of the nest-building *Lasioglossum* (*Dialictus*) species present at our St. Catharines sites.

## Discussion

Observed morphospecies richness of the entire 10-year sample from the St. Catharines sites (147 species) effectively matched that predicted by the abundance-based coverage (ACE, 147 species) and Chao1 estimators (mean  $\pm$  SD =  $145 \pm 9.6$ ) based on combined pan-trap, sweep-net, and flower-net collection data from 2003 (Richards *et al.* 2011). Only three species present in Port Colborne and Wainfleet were entirely absent in samples from St. Catharines; these were *Lasioglossum oblongum* (Lovell) (Hymenoptera: Halictidae), *Lasioglossum pilosum* (Smith) (Hymenoptera: Halictidae), and *Osmia georgica* Cresson (Hymenoptera: Megachilidae). We collected far more species and morphospecies in St. Catharines (147) than in Port Colborne (64) and Wainfleet (61) (Table 1), which was expected since St. Catharines samples were collected for 10 years and in relatively undisturbed as well as regenerating sites.

The current list undoubtedly reflects some biases in the sampling protocols used, and there are certain taxa that appear to be underrepresented in terms of diversity, or should be present given records in areas neighbouring the Niagara Peninsula but are entirely absent

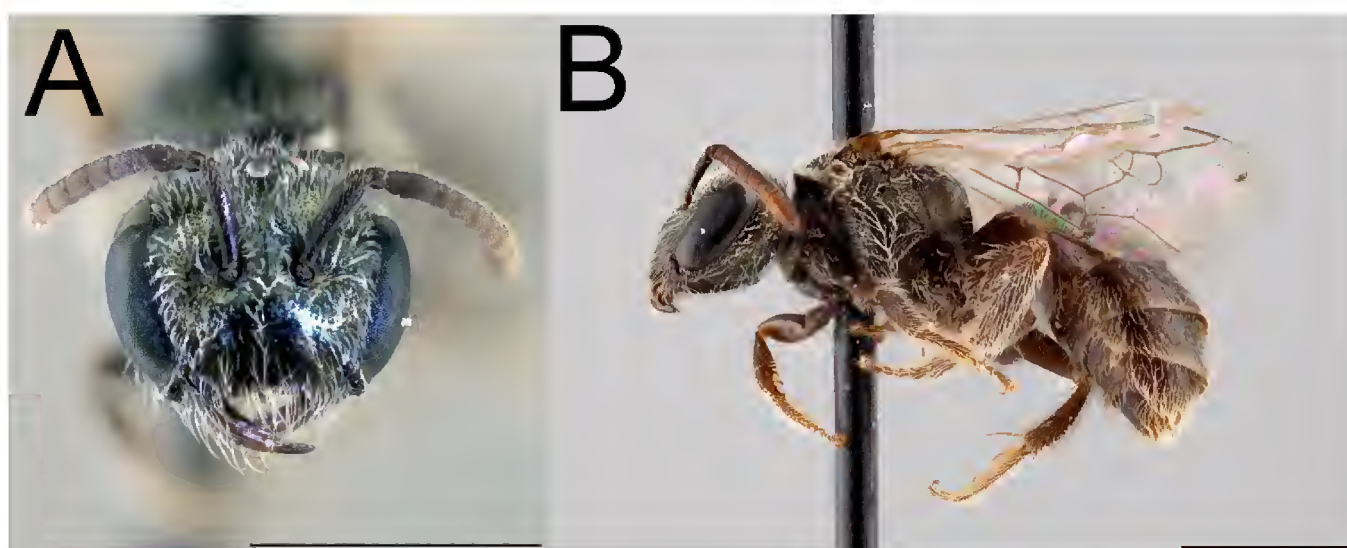


FIGURE 1: Face (A) and lateral view (B) of a female specimen of *Lasioglossum furunculum* Gibbs collected in southern St. Catharines in 2009. Note the absence of a preapical tooth on the mandible and narrow gena in lateral view. Scale bars = 1 mm.



from our samples. We never sampled from trees or tall shrubs, so the species presented here likely reflect a large subsample of the resident bee communities that forages at or near ground level. This might explain the comparatively low diversity of the genus *Andrena*, which frequently visit spring blooming trees and include multiple specialists of willows and other spring blooming plants. Given that pan-trapping was the main sampling method employed among years, it is not surprising that in our samples *Colletes* Latreille (Hymenoptera: Colletidae), a genus usually sampled very well in nets, not pans (Wilson *et al.* 2008), were low in both number and diversity. Forty bee genera occur in southern Ontario (Packer *et al.* 2007; Gibbs *et al.* 2014), and several of these (*Dieunomia* Cockerell (Hymenoptera: Halictidae), *Dianthidium* Cockerell (Hymenoptera: Megachilidae), *Paranthidium* Cockerell and Cockerell (Hymenoptera: Megachilidae), *Epeoloides* Giraud (Hymenoptera: Apidae), *Macropis* Panzer (Hymenoptera: Melittidae), and *Svastra* Holmberg (Hymenoptera: Apidae)) are too rare or transient to be expected in our sites. The melittid genus *Macropis* ranges from Nova Scotia to Washington and south to Georgia (Hurd 1979) and collects floral oils from loosestrife (*Lysimachia*) Linnaeus (Primulaceae) (Cane *et al.* 1983), which was rare in our study sites. Some bees like *Peponapis pruinosa* (Say) (Hymenoptera: Apidae) are expected to be relatively common in the Niagara Region, but this species is restricted to areas where cultivated cucurbits, *Cucurbita* Linnaeus (Cucurbitaceae), are present. *Perdita* Smith (Hymenoptera: Andrenidae), another genus that should be present in our region, is largely composed of small, floral specialists. *Holcopasites* Ashmead (Hymenoptera: Apidae), absent from our collections, are small cleptoparasites of *Calliopsis*; the latter was uncommon in our sites. *Epeolus* Latreille (Hymenoptera: Apidae), a genus of cleptoparasites of *Colletes*, was absent from our samples, although over half a dozen species are known from southern Ontario, including two species recorded from Port Colborne (Romankova 2004). Cleptoparasite and social parasite diversity overall may have been underrepresented in our collections. Our main method of sampling, pan trapping, likely biases collection toward small sweat bees (Halictidae), and underrepresents parasitic species, which spend more time searching for host nests than foraging, and larger bee species that can crawl out of pan traps should they fall in inadvertently (Cane *et al.* 2000; Wilson *et al.* 2008). A study by Cane *et al.* 2000 demonstrated that pan traps failed to catch most species of floral specialists associated with the creosote-bush, *Larrea tridentata* (DC.) Coville (Zygophyllaceae), below which the traps were set. The few specialist species present in our collections were mainly sampled from flowers or sweeps through vegetation.

The number of species found in the present study is lower than that known from the Caledon Hills, located north and east of the Niagara Escarpment and close to the eastern limit of the Carolinian Zone in Ontario. Between two surveys there, one by MacKay and Knerer (1979) in 1968–1969 and another by Grixti and Packer (2006) in 2002–2003, 165 species were recorded, excluding honey bee, *Apis mellifera* Linnaeus (Apidae), and bumble bees, *Bombus* Latreille (Apidae), which were not sampled. Bee surveys taken between 1957 and 1984 at an abandoned field bordering forests comprised of oak, *Quercus* Linnaeus (Fagaceae) and hickory, *Carya* Nuttall (Juglandaceae), (also within the eastern deciduous-Carolinian forest region) at the Edwin S. George Reserve in Livingston County, Michigan, United States of America, yielded a similar number of species (172) (Evans 1986). Given the longer species lists from these similar studies and factors related to sampling, it is likely that at least some additional species occur within or near our study areas, and still more

within the greater Niagara Region.

Exotic species ranged from well-established introductions such as *A. mellifera* and *Megachile rotundata* (Fabricius) (Hymenoptera: Megachilidae) to more recent colonists (Table 1). The Palaearctic leafcutter bee *Megachile ericetorum* Lepeletier (Hymenoptera: Megachilidae), first discovered in the New World in St. Catharines in 2003 (Sheffield *et al.* 2010), is now well-established in the Niagara Region, based on subsequent captures of more than a dozen individuals in St. Catharines in 2006, 2010, 2012, and 2013 and in Port Colborne in 2012 and 2013; and recently in Rochester, New York, United States of America (Jacobi and Stafford 2012). We also collected two introduced *Hylaeus* species of the subgenus *Spatulariella*. *Hylaeus hyalinatus* Smith (Hymenoptera: Colletidae) was first reported in North America in 2001 (Ascher 2001) and then in St. Catharines almost every year from 2003 (Richards *et al.* 2011) to 2013, and was also found in Wainfleet in 2012. *Hylaeus punctatus* (Brullé) (Hymenoptera: Colletidae) was first recorded in Canada in 2011 by Sheffield *et al.* (2011a), and was subsequently discovered in our St. Catharines samples from the same year. *Anthidium manicatum* (Linnaeus) (Hymenoptera: Megachilidae), which was found in almost every sampling year in St. Catharines, was also found in Pt. Colborne and Wainfleet. We also sampled a related introduced species, *Anthidium oblongatum* (Illiger) (Hymenoptera: Megachilidae), which is Palaearctic in origin and has been in Ontario since at least 2002 (Sheffield *et al.* 2011a). Exotic species established in eastern North America for some time include *Lasioglossum leucozonium* (Schrank) (Hymenoptera: Halictidae), *L. zonulum* (Smith) (Hymenoptera: Halictidae) (our only members of the subgenus *Leuchalictus*) (Giles and Ascher 2006), *Chelostoma rapunculi* (Lepeletier) (Hymenoptera: Megachilidae) (Buck *et al.* 2005), and *Megachile sculpturalis* Smith (Hymenoptera: Megachilidae) (Paiero and Buck 2003). The leafcutter bee *Megachile centuncularis* (Linnaeus) (Hymenoptera: Megachilidae) has traditionally been considered to be a Holarctic species, though now there may be reason to suspect that it is exotic in North America as well (Giles and Ascher 2006; Sheffield *et al.* 2011b). Additional collections after 2003 of some of the abovementioned exotic species in the Niagara Peninsula may be indicative of their establishment within Ontario. Continued surveying within the present study region may be important in detecting future introductions as southern Ontario seems to have the highest number of introduced bee species in Canada (16 out of 17 exotic species in Canada (Sheffield *et al.* 2011b)), with one first record for North America of an Old World species discovered in St. Catharines.

Our 10 years of collections represent the most extensive survey of the bee fauna in the Niagara Peninsula to date, and to our knowledge this is the longest continuous survey of any regional bee fauna in Canada. Although rare, transient, or extremely localized species may be discovered in the future, the current list likely encompasses the majority of common species present within the three sampled municipalities. To better detect the regional distribution patterns of bees, comprehensive sampling at additional localities is needed.

## Acknowledgements

In addition to the taxonomic experts mentioned in the Methods who identified a large number of bees and verified many of our designations, we thank Rodrigo León Cordero, Jessi de Haan, and Konrad Karolak for help in preliminary taxonomic sorting of



specimens. This study was made possible with funding and other means of support from the Canadian Pollinator Initiative (CANPOLIN) strategic network, funded by the Natural Sciences and Engineering Research Council (NSERC). Lastly, we thank two anonymous reviewers and an associate editor for their suggestions to improve the manuscript.

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# REDESCRIPTION OF *ANAPHES ATOMARIUS* (BRÈTHES) (HYMENOPTERA: MYMARIDAE) AND COMPARISON WITH SIMILAR SPECIES IN EUROPE AND NORTH AMERICA

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## Abstract

*J. ent. Soc. Ont.* 146: 23–39

*Anaphes atomarius* (Brèthes) (Hymenoptera: Mymaridae) is redescribed based on the holotype and specimens reared from *Listronotus bonariensis* (Kuschel) (Coleoptera: Curculionidae) in Brazil that are assumed tentatively to be conspecific with the type. *Anaphes archettii* Ghidini from Italy is also redescribed, a lectotype designated, and both species are compared to *A. listronoti* Huber and *A. victus* Huber from North America.

*Published November 2015*

## Introduction

The Argentine stem weevil, *Listronotus bonariensis* (Kuschel) (Coleoptera: Curculionidae) is native to South America. It was accidentally introduced into New Zealand where it was discovered in 1927 (Dymock 1989) and has become a major economic pest (Timlin 1964). A search for potential biological control agents was begun by staff at the Commonwealth Institute of Biological Control, South American Station, San Carlos de Bariloche, Argentina, and an egg parasitoid was found and identified as *Anaphes atomarius* (Brèthes) (Hymenoptera: Mymaridae). In 1966 and 1967, consignments of parasitized eggs were sent to New Zealand (Clausen 1977) and specimens were released at Nelson, Lincoln (Canterbury) and Waikato but the species failed to become established as a result of not being able to overwinter (Ferguson *et al.* 2007). Ahmad (1977, 1978) detailed the rearing technique for *L. bonariensis* and the egg parasitoid. Because *L. bonariensis* may occur as a contaminant in grain shipments from New Zealand or elsewhere it is listed as a quarantine pest of pasture grasses and cereals in the European Union (Ostoja-Starzewski 2011). The original 5-line Latin description and sketchy line drawings of wings and antenna are inadequate to define *Anaphes atomarius* and because of its potential for biological control the species is redescribed here, based on the holotype and several other specimens reared from *L. bonariensis* in Brazil. It is compared with similar species reared from known hosts in Europe and North America.

## Methods

Non-type specimens were slide mounted in Canada balsam using the method described by Noyes (1990). Photographs of slide preparations were taken with a ProgRes C14<sup>plus</sup> digital camera attached to a Nikon Eclipse E800 compound microscope, and the resulting layers combined electronically using Auto-Montage<sup>®</sup> (Synoptics Group, Cambridge) or Zerene Stacker<sup>™</sup> (<http://zerenestacker.com>) and, except for primary types, retouched as needed with Adobe<sup>®</sup> Photoshop (Adobe Systems for Windows). Measurements of morphological structures are given in micrometres (µm), following Huber (1992, 2006). Abbreviations used are: fl<sub>x</sub> = funicle or flagellar segment, mps = multiporous plate sensillum. Specimens are deposited in the following institutions.

CNC – Canada, Ontario, Ottawa, Canadian National Collection of Insects.

DEZA – Italy, Naples, Portici, Dipartimento di Entomologia e Zoologia Agraria dell'Università degli Studi di Napoli «Frederico II».

MACN – Argentina, Buenos Aires, División Entomología, Museo Argentino de Ciencias Naturales “Bernardino Rivadavia”.

### *Anaphes atomarius* (Brèthes)

*Anaphoidea atomaria* Brèthes, 1913: 100 (original description).

*Patasson atomarius*: Ogloblin, 1964: 39 (generic transfer).

*Patasson atomarius*: De Santis, 1967: 109 (catalogue).

*Patasson atomarius*: Clausen, 1977: 272 (host, biological control).

*Patasson atomaria*: De Santis, 1979: 371 (catalogue).

*Patasson atomarius*: Ahmad, 1977: 151 (host, percent parasitism).

*Patasson atomarius*: Ahmad, 1978: 161 (laboratory rearing, longevity).

*Patasson atomarium*: Dymock, 1989: 23 (biological control).

*Anaphes atomarius*: Huber, 1992: 72 (list, implied generic transfer).

**Type material.** Holotype ♀ (MACN), on slide (Fig. 2) labelled: 1. “Patasson atomarius ♀ Brèthes]. Det. A. Ogloblin”. 2. “A 14”. 3. “Anaphoidea atomaria Br. 10545”. Some illegible letters in faded ink and the number 53 in pencil are also on the labels.

Type locality: the original description gives the type locality and collecting date as General Urquiza and 1.xi.1912. The locality is now in Villa Urquiza, an area in greater Buenos Aires.

**Other Material Examined.** BRAZIL. Rio Grande do Sul: Passo Fundo, 14.viii.1985, D.N Gassen, ex. *L. bonariensis* (1♀ and 4♂, CNC).

**Diagnosis.** *Anaphes atomarius* belongs to a complex of species with 2 mps on fl<sub>2</sub> of each antenna in females. The holotype differs from *A. archettii* (described below) and *Anaphes listronoti* Huber by the fore wing with double line of setae separating the medial space from the posterior margin of the wing (a single line in *A. listronoti*), and narrower fore wing.

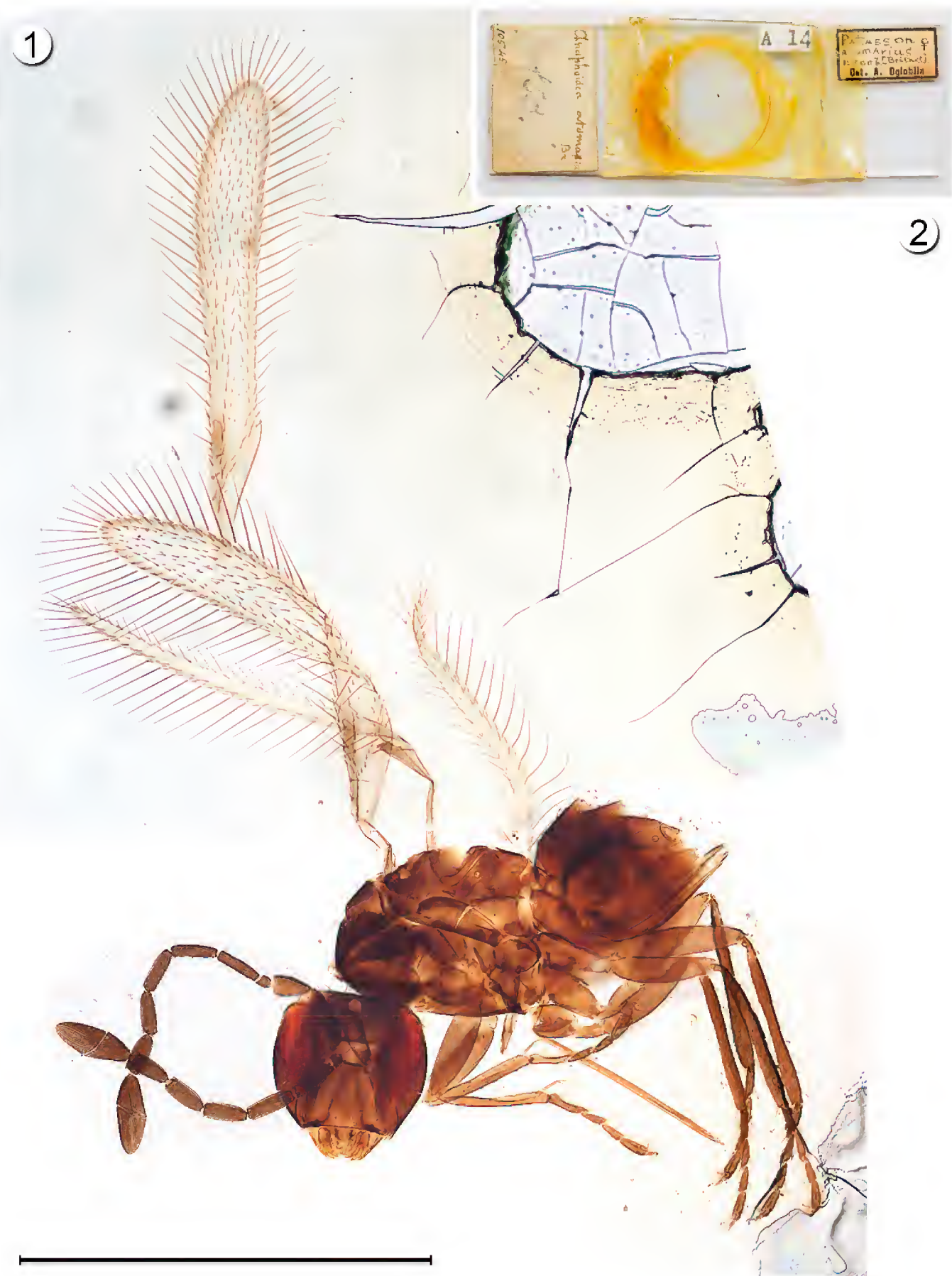
Incidentally, the line drawing of the fore wing of the holotype of *A. listronoti* (Huber *et al.* 1997, fig. 11) differs from its photograph (Huber 2006, fig. 23) in that the setal line between the medial space and posterior margin appears partly double in the former but single in the latter. I rechecked the holotype and the photograph showing a single setal line is correct. *Anaphes atomarius* differs from *A. victus* Huber by the narrower fore wing, with length to width ratio at least 8.0 (at most 6.7 in *A. victus*). The reared female from Brazil that I tentatively identify as *A. atomarius* has the fore wing with a single setal line separating medial space from hind margin and a slightly wider wing (length to width ratio of 7.37).

**Description. Female.** Holotype (Fig. 1) body length 445 (mesosoma + metasoma only) (total length including head = 500 in original description). **Head.** Head width 189. **Antenna.** Length to width ratio of segments: scape + radicle 79/25 (3.16), pedicel 49/29 (1.69), fl<sub>1</sub> 21/14 (1.5), fl<sub>2</sub> 57/19 (3.00), fl<sub>3</sub> 57/21 (2.71), fl<sub>4</sub> 57/22 (2.59), fl<sub>5</sub> 56/21 (2.67), fl<sub>6</sub> 52/22 (2.36), club 100/38 (2.63); fl<sub>2</sub>–fl<sub>6</sub> each with 2 mps (Fig. 3). **Wings.** Fore wing (Fig. 4) length to width ratio 620/77 (8.05); longest marginal setae about 122, marginal space length 62, with double line of setae separating marginal space from hind margin (Fig. 4). Hind wing length 394, width 23, longest marginal setae about 109. **Legs.** Metatibia length 214, metatarsomere 1–4 lengths 32, 38, 34, 31; metatasomere 1  $0.84 \times$  length of metatarsomere 2. **Metasoma.** Ovipositor sheath length 277, extending under mesosoma to about level of anterior margin of mesocoxa (Fig. 5) and slightly exerted posteriorly (Fig. 6); ovipositor length to metatibia length ratio 1.29.

**Reared female specimen from Brazil.** Body length 490 (mesosoma + metasoma only). **Head.** Head (Fig. 7) width 193. **Antenna.** Scape with faint oblique striations on inner surface (Figs 7, 8). Length to width ratio of antennal articles: scape + radicle 107/24 (4.46), pedicel 49/28 (1.75), fl<sub>1</sub> 26/16 (1.63), fl<sub>2</sub> 64/17 (3.76), fl<sub>3</sub> 64/17 (3.76), fl<sub>4</sub> 62/16 (3.88), fl<sub>5</sub> 62/18 (3.44), fl<sub>6</sub> 58/20 (2.90), club 104/37 (2.81); fl<sub>2</sub>–fl<sub>6</sub> each with 2 mps (Fig. 8). **Mesosoma.** Scutellum (Fig. 9) with campaniform sensilla separated by  $3.2 \times$  their diameter. **Wings.** Fore wing length to width ratio 656/89 (7.37); longest marginal setae about 127, marginal space length 101, with single line of setae separating marginal space from hind margin. Hind wing length to width ratio (for a male specimen) 642/29. **Legs.** Metatibia length 208, metatarsomere 1–4 lengths 33, 39, 40, 35; metatasomere 1  $0.85 \times$  length of metatarsomere 2. **Metasoma.** Gaster (Fig. 10) about  $0.9 \times$  as long as mesosoma. Ovipositor length 294, extending under mesosoma to about level of anterior margin of mesocoxa (Fig. 11); ovipositor length to metatibia length ratio 1.41.

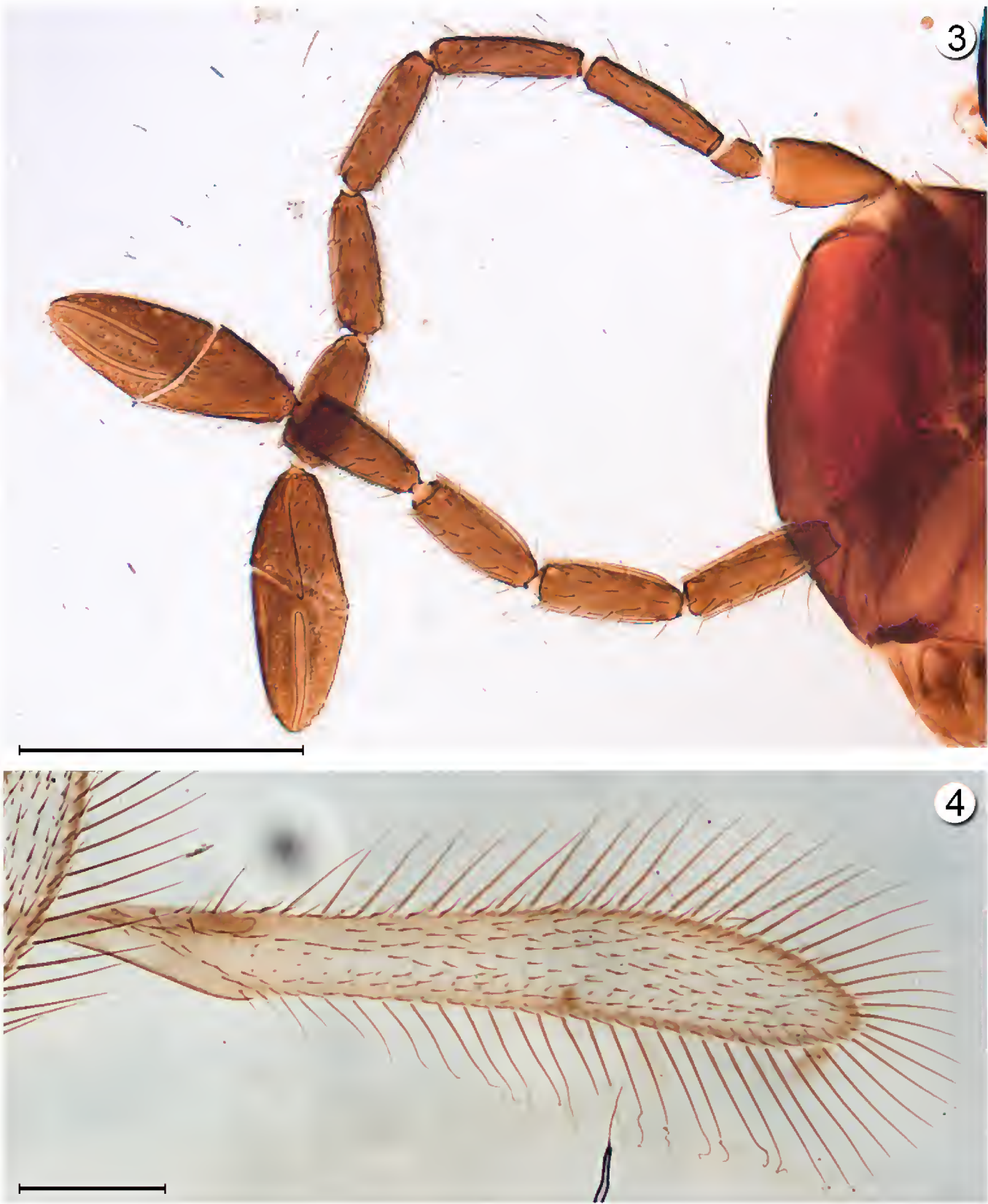
**Reared male specimens from Brazil.** Body length (n=1, on slide) 645. **Head** as in Figs 12 and 13. **Antenna.** Length of segments (n=3) (Fig. 14): scape + radicle 91–97, pedicel 48, fl<sub>1</sub> 4–5, fl<sub>2</sub> 76–81, fl<sub>3</sub> 85–86, fl<sub>4</sub> 81–83, fl<sub>5</sub> 80–82, fl<sub>6</sub> 78–79, fl<sub>7</sub> 76–82, fl<sub>8</sub> 76–78, fl<sub>9</sub> 80–84, fl<sub>10</sub> 76–80, fl<sub>11</sub> 77–82. Length/width of fl<sub>5</sub> 3.75–4.04. Total flagellum length 797–815. **Mesosoma.** As in Fig 16. **Wings.** As in Fig 15. **Metasoma.** Gaster (Fig. 17) slightly longer than high. Genitalia as in Fig. 18 (and see comments in Discussion). The four males are assumed to be conspecific with the reared female based on being obtained from the same rearing event.



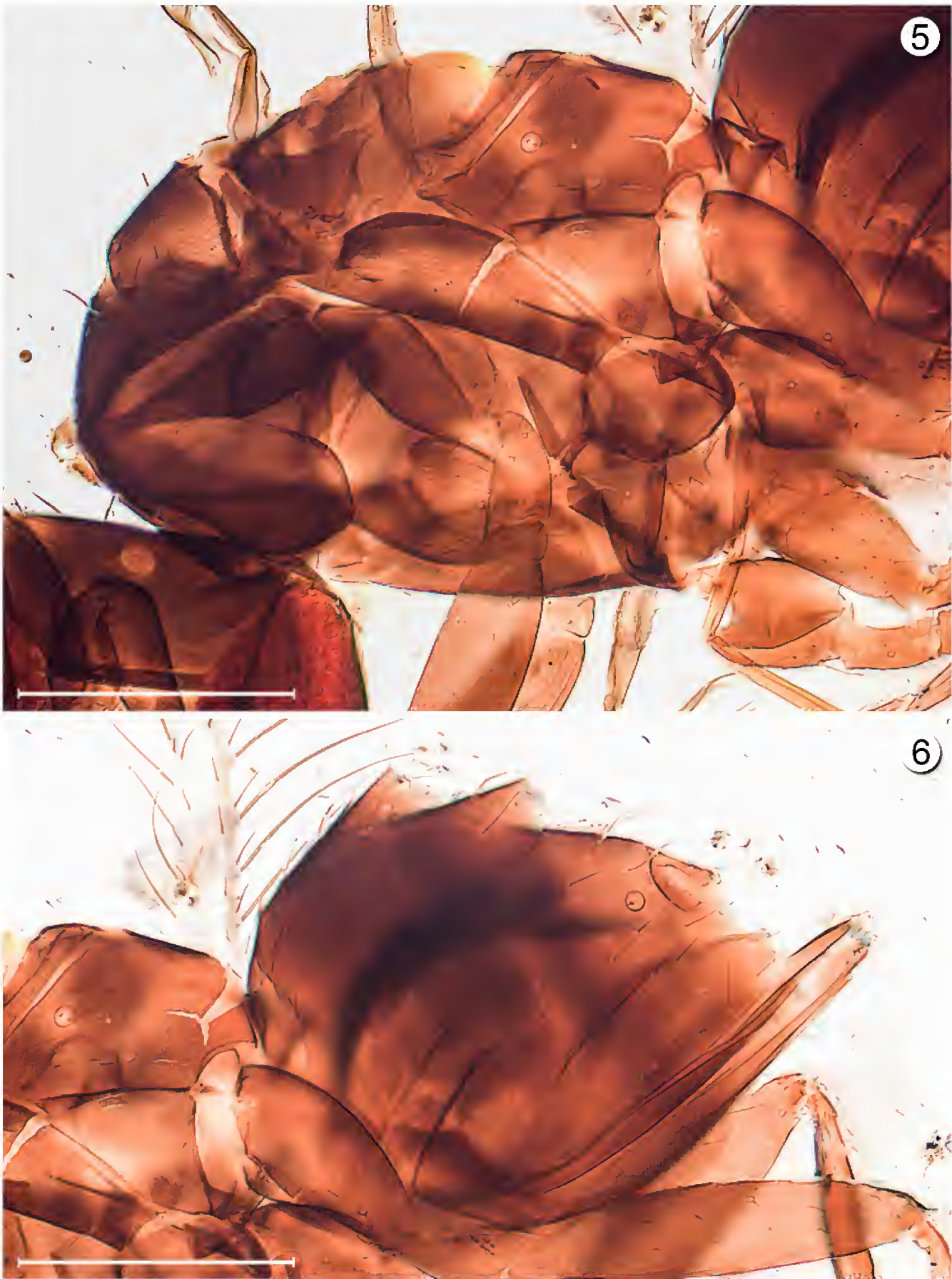


FIGURES 1–2. *Anaphoidea atomaria*, holotype. 1, habitus; 2, type slide. Scale bar = 500  $\mu\text{m}$ .



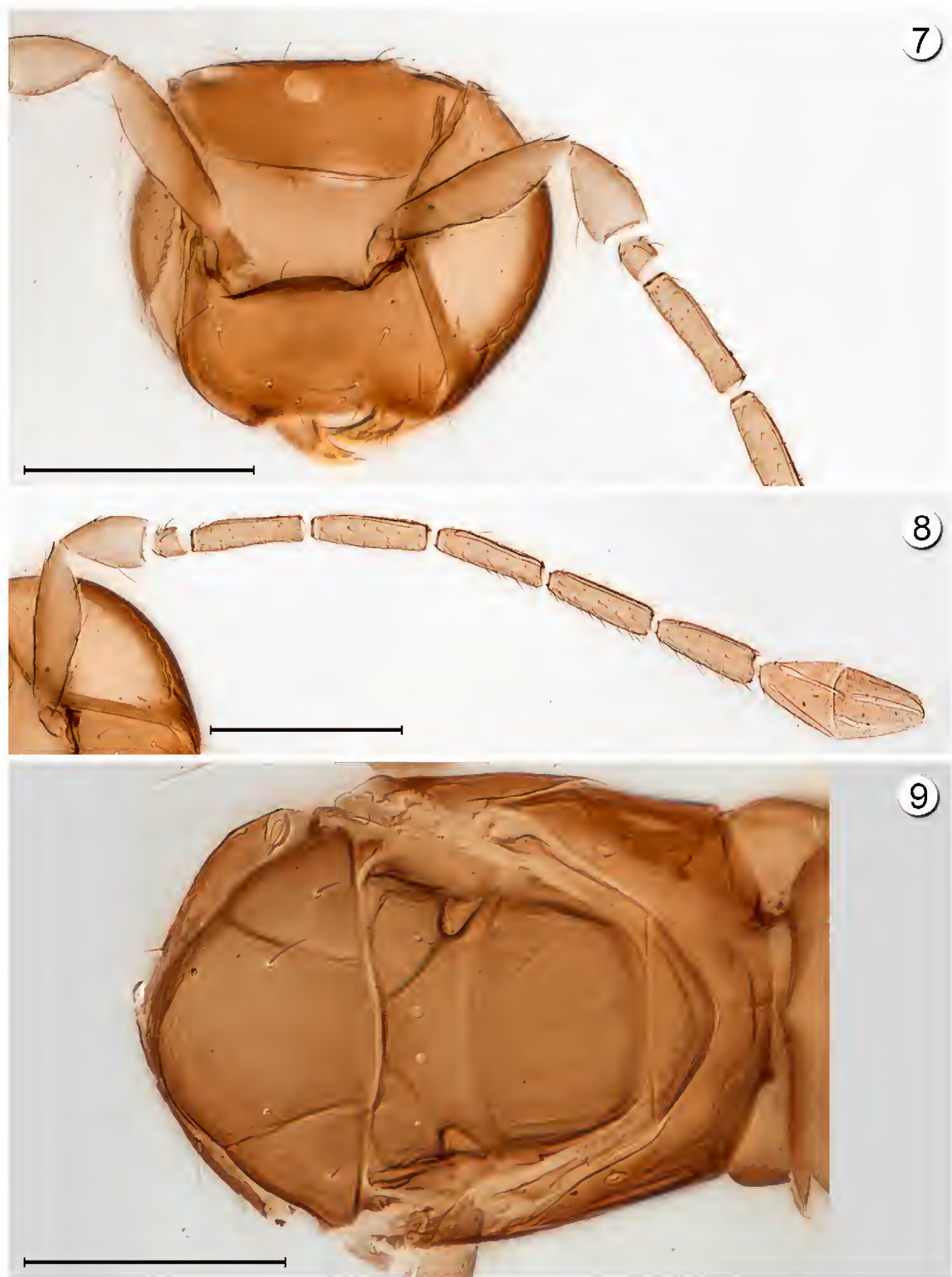


FIGURES 3–4. *Anaphoidea atomaria*, holotype. 3, antennae; 4, fore wing. Scale bars = 100  $\mu\text{m}$ .

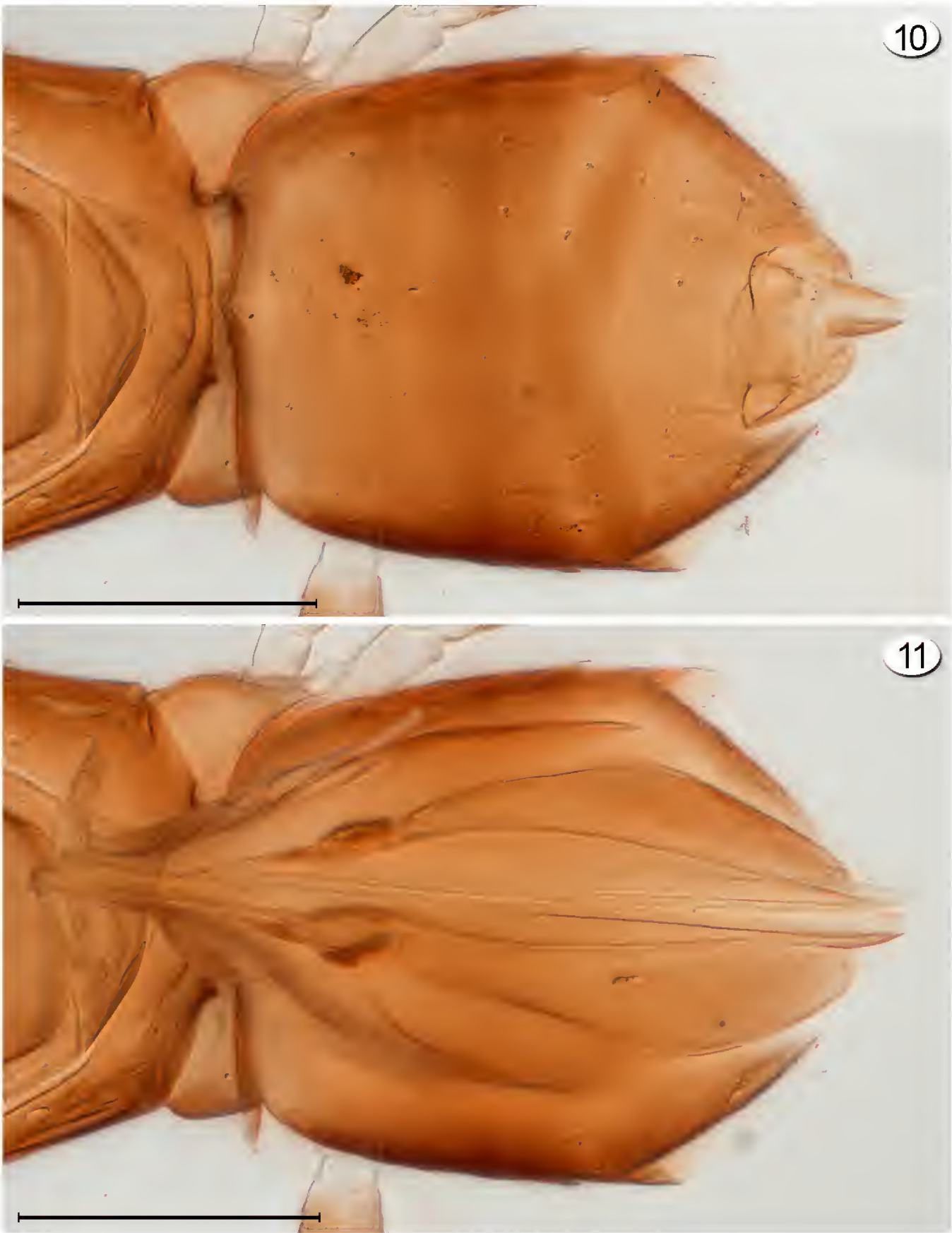


FIGURES 5–6. *Anaphoidea atomaria*, holotype. 5, mesosoma, lateral; 6, metasoma, lateral. Scale bars = 100  $\mu$ m.



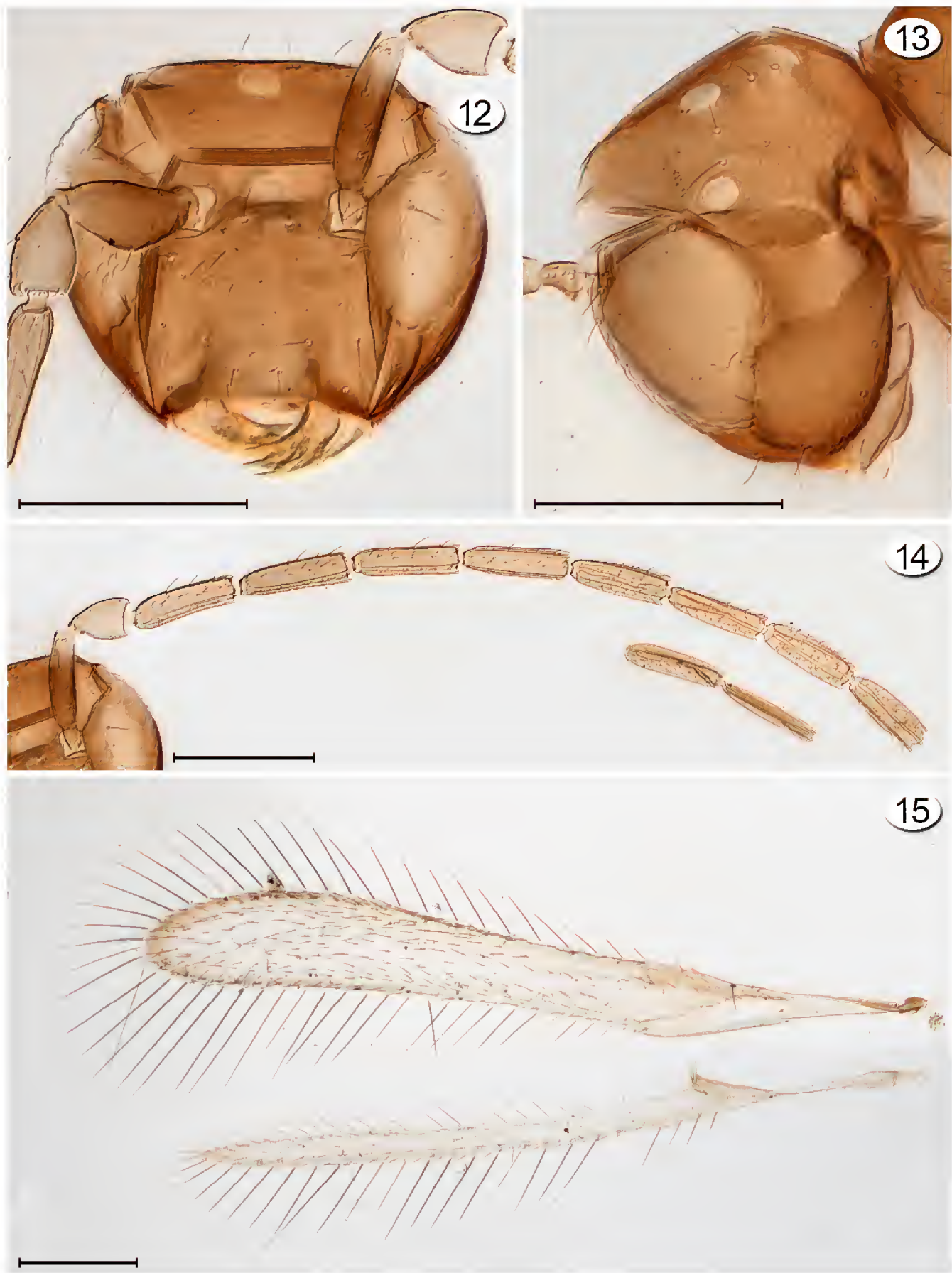


FIGURES 7–9. *Anaphes ?atomarius*, reared female from Brazil. 7, head, anterior; 8, antenna; 9, mesosoma, dorsal. Scale bars = 100  $\mu$ m.

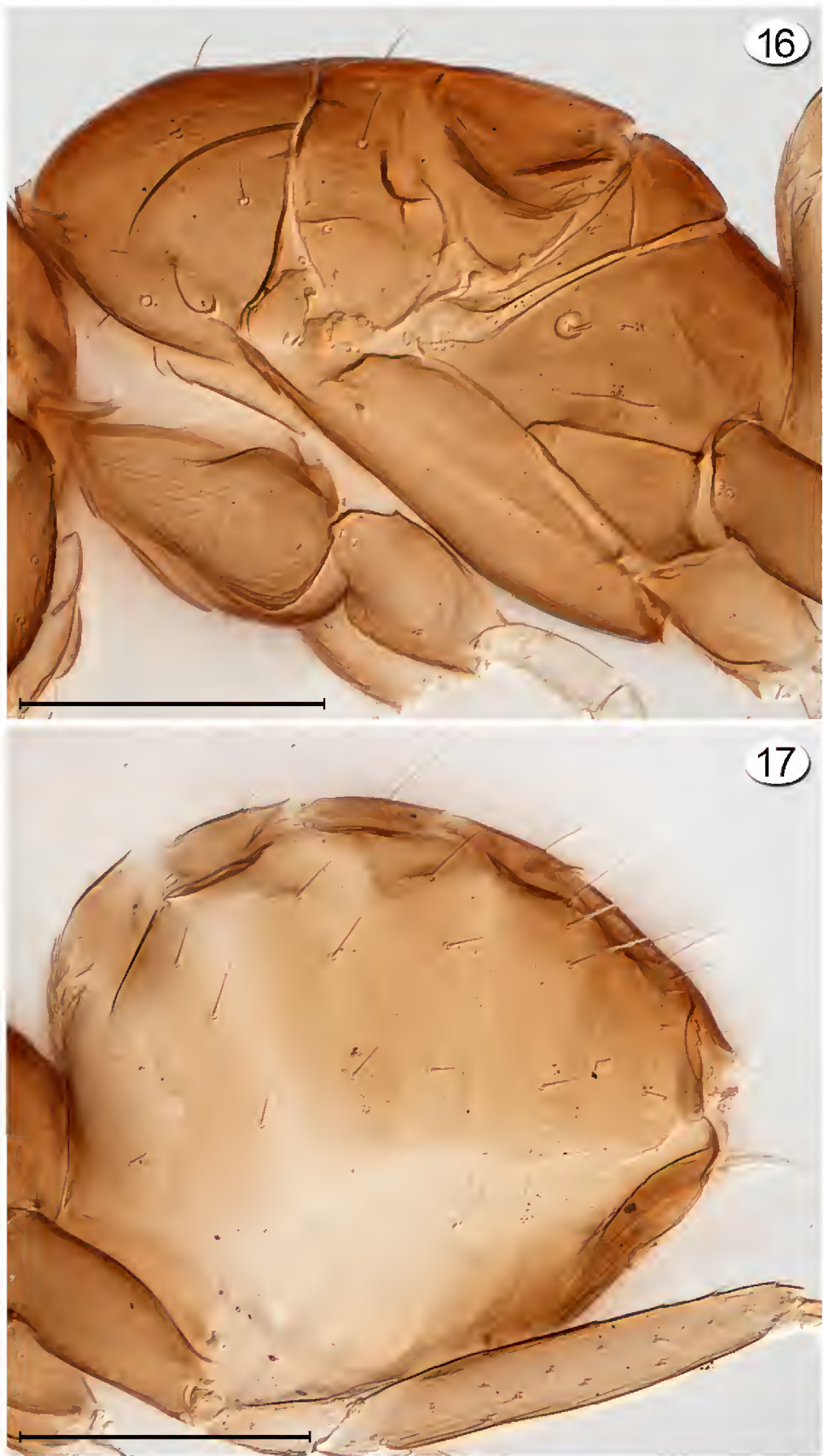


FIGURES 10–11. *Anaphes* ?*atomarius*, reared female from Brazil, apex of mesosoma + metasoma; 10, dorsal surface; 11, ovipositor, dorsal view (as seen through metasoma). Scale bars = 100 μm.





FIGURES 12–15. *Anaphes* ?*atomarius*, reared male from Brazil. 12, head, anterior; 13, head, dorsolateral; 14, antenna; 15, wings. Scale bars = 100  $\mu$ m.



FIGURES 16–17. *Anaphes atomarius*, reared male from Brazil, lateral. 16, mesosoma; 17, metasoma. Scale bars = 100 µm.





FIGURE 18. *Anaphes* ?*atomarius*, reared male from Brazil, dorsal, genitalia. Scale bar = 100  $\mu$ m.

### *Anaphes archettii* Ghidini

*Anaphes archettii* Ghidini, 1945: 39 (original description).

*Anaphes archettii*: Viggiani and Jesu, 1988: 1020 (host cited).

*Anaphes archettii*: Huber, 1992: 72 (list).

*Anaphes archettii*: Viggiani, 1994: 472 (male genitalia).

*Anaphes archettii*: Pagliano and Navone, 1995: 36 (list).

*Anaphes archettii*: Jesu, 2002: 111 (host cited).

*Anaphes archettii*: Pintureau, 2012: 33 (list).

**Type material.** Lectotype ♀, here designated (DEZA), on slide (Fig. 20) labelled 1. Littoria, 13.v.1943 ex *Lixus junci* coll. F.M Ghidini". 2. "Lectotype ♀ des. Huber 2014". 3. "*Anaphes archettii* Ghidini 2♀". 4. "Paralectotype ♀ *Anaphes archettii*". Type locality: Italy, Lazio, Agro Pontino [a plain in Latina Province south and southeast of the provincial capital, Latina). The former name of Latina was Littoria (used in the original description).

Paralectotypes. 1♀ and 1♂ (DEZA), with same data as lectotype. The female paralectotype is on the same slide as the lectotype, the male on another slide; both were examined. Three other specimens (DEZA) remain from the original series

but are in poor condition; they were not examined. All other original specimens are lost (G. Viggiani, personal communication).

**Diagnosis.** *Anaphes archettii* belongs to the same species complex as *A. atomarius*. It differs from *A. atomarius* by four features: 1) longer body length (at least 770 long vs 500 in *A. atomarius* holotype), 2) fore wing with a single line of setae separating the medial space from the posterior margin (double line in *atomarius* holotype), 3) fore wing length to width ratio 5.39 (8.05 in *atomarius* holotype) and 4) ovipositor to metatibia length ratio 1.77 (1.29 in *A. atomarius*). The body length of *A. archettii* is at least 770, based on Ghidini (1945) compared to at most 693 in *A. victus* and 723 in *A. listronoti*.

**Description. Female.** Lectotype (Fig. 19) body length (mesosoma + metasoma only) 792 (total length including head = 770–850 in original description). **Head.** Head width 314. **Antenna.** Length to width ratio of segments (scape–fl<sub>5</sub> from paralectotype): scape + radicle 155/47, pedicel 65/38, fl<sub>1</sub> 36/19, fl<sub>2</sub> 101/26, fl<sub>3</sub> 101/27, fl<sub>4</sub> 99/30, fl<sub>5</sub> 91/29, fl<sub>6</sub> 90/30, clava 145/45; fl<sub>2</sub>–fl<sub>6</sub> each with 2 mps (Figs 21, 22). **Wings.** Fore wing (Fig. 26 [male]) length to width ratio 1013/188 (5.39); longest marginal setae about 150, marginal space length 146, with single line of setae separating marginal space from hind margin. Hind wing length 904, width 58, longest marginal setae about 130. **Legs.** Metatibia length (paralectotype) 334, metatarsomere 1–4 lengths 61, 66, 58, 35; metatarsomere 1  $0.92 \times$  length of metatarsomere 2. **Metasoma.** Ovipositor sheath length 592, extending under mesosoma to about level of anterior margin of mesocoxa (Fig. 23); ovipositor length to metatibia length ratio 1.77.

**Male.** Body length (from original description) 0.65–0.70 mm. **Antenna.** Length of segments (Fig. 24) (paralectotype): scape + radicle 128/40, pedicel 51/39, fl<sub>1</sub> 9, fl<sub>2</sub> 125, fl<sub>3</sub> 119, fl<sub>4</sub> 118, fl<sub>5</sub> 119/23, fl<sub>6</sub> 116, fl<sub>7</sub> 116, fl<sub>8</sub> 115, fl<sub>9</sub> 111, fl<sub>10</sub> 109, fl<sub>11</sub> 114. Length to width ratio of fl<sub>5</sub> 5.04. Total flagellum length 1171. Fore wing as in Fig. 26. Genitalia as in Fig. 25 (and see comments in Discussion).

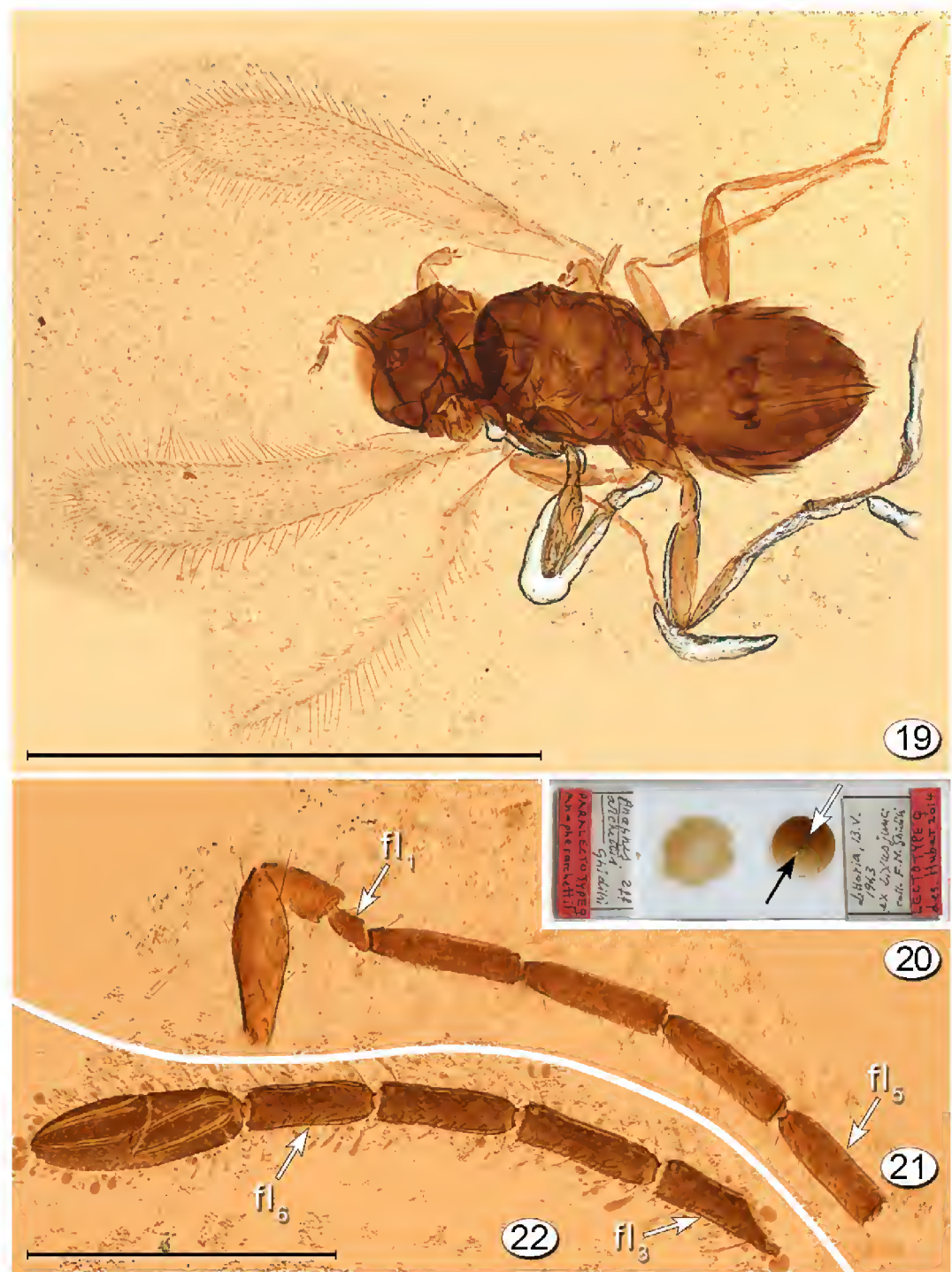
## Discussion

Only four species of *Anaphes* have been described from South America: three in *A. (Yungaburra)* and one, *A. atomarius*, in *A. (Anaphes)* (Huber 1992). *Anaphes atomarius* belongs to the *crassicornis* species group, in which the clava is 2-segmented. Among species described from the Western Hemisphere *A. atomarius* would key to couplet 12 in Huber (2006), which leads to *A. listronoti*, *A. sordidatus* (Girault) and *A. victus*. *Anaphes victus* and some specimens of *A. listronotus* Huber were reared from *Listronotus oregonensis* (LeConte) (Coleoptera: Curculionidae) among other species, and *A. sordidatus* was reared from *Tyloderma foveolatum* (LeConte) (Coleoptera: Curculionidae). Specimens of all three species sometimes or always have 2 mps on fl<sub>2</sub> of the female antenna, in contrast to other *Anaphes* species that have at most 1 or, usually, 0 mps on fl<sub>2</sub>. Several Old World (European) species also have 2 mps on fl<sub>2</sub>, but only one of them, *A. archettii*, is treated here for comparison with *A. atomarius* because the types were reared from a known host.

The specimens from Passo Fundo, about 900 km from the type locality of *A. atomarius*, match the holotype fairly well but not perfectly. I tentatively treat the differences as intraspecific variation until shown otherwise by further rearing and morphological study



of additional specimens reared from *L. bonariensis*, preferably from nearer the type locality. Because of the slight morphological differences, the species name *atomarius* may not be correctly applied to the reared specimens I examined. Regarding specimens introduced into New Zealand, it is not known who made the species identification, whether voucher specimens from the releases or studies were kept or, if so, where they are deposited. Therefore their identity cannot be checked. Because no voucher specimens were located from previous



FIGURES 19–22. *Anaphes archettii*, lectotype. 19, habitus; 20, type slide, 21, paralectotype antenna, from scape (radicle missing) to fl<sub>5</sub>; 22, lectotype antenna, from fl<sub>3</sub> (part) to clava. Scale bars: 19 = 1000 µm, 21 and 22 = 200 µm.



FIGURES 23–26. *Anaphes archettii*, types. 23, female paralectotype, body dorsal; 24, male paralectotype, head + antenna; 25, male genitalia, lateral; 26, fore wing. Scale bars = 200  $\mu\text{m}$ .



publications that use the name *A. atomarius* I cannot be sure whether the species name was correctly applied in those publications either. Like most species of *Anaphes*, the holotype of *A. atomarius* was not reared so its host is unknown. It would be expedient to assume that the name *A. atomarius* was correctly applied to all specimens reared from *L. bonariensis* because then the name would be associated with specimens reared from a known host that happens also to be a pest of economic importance. But this cannot be done until more evidence of conspecificity is obtained. That may be impossible because the holotype is slide mounted so other lines of evidence such as DNA barcoding or biological information cannot be obtained from it for comparison with freshly reared specimens from known hosts.

The possibility exists that a complex of similar *Anaphes* species in South America uses *L. bonariensis* as a host, just as a complex of species exists on *L. oregonensis* in North America. Species in the latter complex differ in biology, e.g., in the number of individuals reared from a single host egg of *L. oregonensis*—*A. listronotus* is gregarious and *A. victus* is solitary (Huber *et al.* 1997). Unfortunately, publications on the biology of *A. atomarius* do not state how many adults emerge from a single host egg and this information was not recorded in the five reared specimens in this study. Another possibility is that *A. atomarius* is the same as one of the North American species. The fact that one species was described from Brazil and the others from Canada or the United States of America is not a problem because various species of Mymaridae in the Western Hemisphere are known to have wide distributions that extend from Canada, or at least somewhere north of Mexico, to Argentina. Additional rearing is needed of *A. 'atomarius'* from *Listronotus* spp. in South America for detailed morphological study and, if colonies can be established, laboratory crossing experiments with the North American species, preferably with the addition of molecular evidence to see if species are the same or different.

Ghidini (1945) reared numerous specimens of *A. archettii* from *Lixus junci* Boheman (Coleoptera: Curculionidae) on sugar beet (*Beta vulgaris* Linnaeus) (Chenopodiaceae) in Italy but did not state how many emerged from a single weevil egg. Apart from the specimens discussed above, the original material is lost (Viggiani, personal communication). Viggiani (1994) illustrated the male genitalia (photographed in Fig. 25) and showed that various *Anaphes* species could be distinguished by measurements of various genitalic parts. The problem is that association of males with females is only certain for the few *Anaphes* species reared from economically important hosts, whereas descriptions of most *Anaphes* species are based on females only, the corresponding males being unknown or not certainly associated. Because the genitalia of only three males of *A. atomarius* from Brazil and one of *A. archettii* are available for study little can be said about variation. In any case, there appears to be no difference in measurements.

Body length in the four *Anaphes* species discussed above may be correlated with host egg size and number of individuals developing in a single egg. The gregarious or solitary nature of *A. atomarius* and *A. archettii* must first be determined, however. A host for each of the four species is known if one expediently, but perhaps incorrectly, assumes that specimens reared from *L. bonariensis* are indeed *A. atomarius*. If eggs of *L. junci* are larger than those of any of the *Listronotus* species that may account for the larger body size of *A. archettii* compared to the other *Anaphes* species. It would be interesting to obtain living *A. archettii* from *L. junci* and try to rear them on *L. oregonensis* in order to determine whether the host range can be extended and, if so, see if specimens reared from *L. oregonensis* are

smaller than when reared on *L. junci*. If they are, then the body length difference proposed above to separate *A. archettii* from *A. listronoti* or *A. victus* does not distinguish these species and other differences need to be found. Ultimately, molecular evidence and cross breeding may be needed to distinguish correctly these (and other) *Anaphes* species. It may show that at least two of them are conspecific.

## Acknowledgements

I thank J. Martinez (MACN) for the loan of the holotype of *A. atomarius* and G. Viggiani (DEZA) for the loan of three syntypes of *A. archettii* and information on the type locality. D. Ward, New Zealand Arthropod Collection and D. Gunawardana, Plant Health and Environment Laboratory, Ministry of Agriculture and Forestry, Auckland, searched for voucher specimens of *A. atomarius* but could not locate any. J. Read (CNC) is gratefully acknowledged for preparing the plate of illustrations.

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# PHEROMONE RACES OF *OSTRINIA NUBILALIS* HÜBNER (LEPIDOPTERA: CRAMBIDAE) INFESTING GRAIN CORN IN MANITOBA, ONTARIO, AND QUÉBEC PROVINCES OF CANADA

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## Abstract

*J. ent. Soc. Ont.* 146: 41–49

*Ostrinia nubilalis* (Hübner) (Lepidoptera: Crambidae), European corn borer, is an economic pest of *Zea mays* (Linnaeus) (Poaceae) and other vegetable crops that is distributed throughout the agricultural production regions of Ontario, Québec, and Manitoba in Canada. Two phenotypic races of *O. nubilalis* have been identified that differ in the proportion of isomers of 11-tetradecenyl acetate (11-14:OAc) in their sex pheromone. The Z-race (Z-11-14:OAc) is the predominant race in the United States of America, known to inhabit *Zea mays* as its primary host, whereas the E-race (E-11-14:OAc) infests a wider host range, including many vegetable crops, and is only found within the Eastern coastal states of the United States of America. Collections of *O. nubilalis* were made from grain corn in agricultural regions of Ontario, Québec, and Manitoba in 1997, 2008, 2009, and 2010, and females were analyzed for pheromone race using gas chromatography (GC). Only Z-race *O. nubilalis* were found in Ontario (from Essex to Leeds and Grenville Counties) and in Southern Manitoba. E-race individuals were detected in collections from Ottawa, Ontario and St. Anicet, Québec, with an increasing proportion of E-race phenotypes in samples from west to east. This is the first report of pheromone race determination using GC among Canadian *O. nubilalis* populations and the first documentation of E-race *O. nubilalis* in Canada using GC.

*Published December 2015*

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## Introduction

*Ostrinia nubilalis* (Hübner) (Lepidoptera: Crambidae), European corn borer, has been an economic pest of corn, *Zea mays* (Linnaeus) Poaceae, throughout North America since introduction early in the 20<sup>th</sup> century (Caffrey and Worthley 1927; Mason *et al.* 1996). Two phenotypic races of this species have been identified that differ in the proportion of 11-tetradecenyl acetate (11-14:OAc) geometrical isomers in their sex pheromone composition (Klun and Brindley 1970). Although *O. nubilalis* is reported to utilize over 200 host plants, it predominantly infests corn, as its common name implies; however, the E-race (E-11-14:OAc) inhabits a wider host range, including peppers *Capsicum* spp. (Linnaeus) Solanaceae, potato *Solanum tuberosum* (Linnaeus) Solanaceae, tomato *Solanum lycopersicum* (Linnaeus) Solanaceae, and wheat *Triticum aestivum* (Linnaeus) Poaceae, as well as corn, whereas the Z-race (Z-11-14:OAc) has a strong fidelity to corn (Bontemps *et al.* 2004; Mason *et al.* 1996). The Z-race is present throughout the North American range of *O. nubilalis* (Palmer *et al.* 1985); populations within the United States of America Corn belt are dominated by the Z-race (Mason *et al.* 1996; Showers *et al.* 1974), whereas the Northeastern coastal states contain greater proportions of the E-race (Klun and Brindley 1970; Mason *et al.* 1996; O'Rourke *et al.* 2010; Roelofs *et al.* 1972; Roelofs *et al.* 1985). Regional pheromone race identification of *O. nubilalis* is important for effective integrated pest management in agricultural crops including population monitoring using pheromone traps (DuRant *et al.* 1995) and for resistance management implications (Bontemps *et al.* 2004; O'Rourke *et al.* 2010). The major corn producing areas in Canada are in southern portions of Manitoba, Ontario, and Québec (Hamel and Dorff 2013); however, the pheromone race composition of *O. nubilalis* from these regions has not been reported.

Klun *et al.* (1975) reported results for captures of *O. nubilalis* males in pheromone traps from several locations in Canada. They tested blends of the two pheromone isomers ranging from dominance of Z at one extreme to dominance of E in the lures at the other end of the spectrum. Their trapping data showed that more than 15% of males trapped at Simcoe, Ontario and St. Jean, Québec were attracted to E pheromone blends out of a total catch of at least 45 moths in each case. At three other locations, moths predominantly were caught in traps baited with Z blends. Although pheromone trapping of males can provide an indication of presence of E and Z races in a population, this method is not definitive because males exhibit different levels of response to E and Z independently baited lures (Glover *et al.* 1991; Mason *et al.* 1997; Pelozuelo and Frérot 2007). McLeod *et al.* (1979) reported that male *O. nubilalis* collected from two Ontario locations and one population from St. Rémi, Québec responded most strongly to Z-11-14:OAc using electroantennograms. However, another population that infested corn later in the same growing season from the Québec location responded with greater affinity to E-11-14:OAc. The most reliable method of race determination is analysis through gas chromatography of excised female pheromone glands or by analysis of race-specific single nucleotide polymorphism (SNP) genetic markers (Coates *et al.* 2013).

Although grain corn is the second largest crop produced in Ontario by acreage, vegetable crops such as field tomatoes, sweet corn, and peppers also provide substantial farm value to the agricultural economy within the province (Hagerman 1997). The presence of significant acreages of fruit and vegetable crops in Essex, Chatham-Kent, and Niagara

Counties in Ontario, which have the potential to support E-race *O. nubilalis*, and reports of infestation of winter wheat *T. aestivum* (Linnaeus) Poaceae in Québec and eastern Ontario (F. Meloche, personal communication) prompted the investigation of the composition of pheromone races in Canadian populations of *O. nubilalis*. Although Klun *et al.* (1975) and McLeod (1979) provided results for males collected in E- and Z-baited traps, the pheromone composition has not been documented with race-specific analysis through gas chromatography or SNP analysis. Although there is some hybridization in the field, E and Z populations are usually isolated due to multiple reproductive barriers (Dopman *et al.* 2010). The present study represents the first report of race-specific testing of Canadian *O. nubilalis* populations using gas chromatography; these results were generated prior to the development and publication of methods for SNP analysis (Coates *et al.* 2013). Populations of *O. nubilalis* were collected from commercial grain corn fields in Ontario, Québec, and Manitoba in 1997, 2008, 2009, and 2010, and sent to C.E.M. at the University of Delaware for pheromone gland analysis of females using gas chromatography.

## Materials and Methods

### Insect Specimens

*O. nubilalis* larvae were collected in September or October of each sampling year from commercial grain corn fields that had not been planted with transgenic hybrids that express *Bacillus thuringiensis* (Berliner) (*Bt*) Bacillales insecticidal proteins (*Bt*-corn) or from non-*Bt* refuge plants within *Bt*-corn fields (Table 1, Fig. 1). In 1997, 50 field-collected larvae from each location were cooled and directly shipped, in cardboard larval rearing rings with artificial diet, to C.E.M. for pheromone analysis. In 2008, 2009, and 2010, corn stalks containing diapausing larvae were removed from growers' fields and kept over winter in a non-heated barn at the University of Guelph Ridgetown Campus (Ridgetown, Ontario). Following termination of diapause, larvae were extracted from the corn stalks and transferred into rearing dishes with cardboard pupation rings, which were placed in growth chambers maintained at 16:8 L:D, 27 °C photoperiod, 18 °C scotoperiod, and 75 % relative humidity (RH) to establish laboratory colonies; original colony sizes ranged from 20–70 individuals. After multiple generations of laboratory rearing (Table 1), pupae were removed from the colony, sexed, and female pupae were shipped to C.E.M. for pheromone analysis.

Upon receipt by C.E.M., individual larvae and/or pupae were housed in 28 ml plastic food service cups containing cotton rolls saturated with water, and these were placed in a growth chamber set on a reversed photoperiod to facilitate gland removal at regular working hours. Through pupation and eclosion, conditions were set at 25 °C, 16:8 (L:D) photoperiod, and 50–80 % RH. Drinking water was provided for newly emerged moths, and females were set aside for pheromone analysis.

Pheromone ring glands of females were excised with micro-scissors at the non-sclerotized terminal segment, just anterior of the single ring gland, during the 7<sup>th</sup> h of scotophase the second day after eclosion (24–48 h old). Each gland was placed into a 50- $\mu$ l point-tipped auto-sampler vial containing 5  $\mu$ l of heptane and an internal standard of 4.5 ng cis-7-tetradecenyl acetate (Z-7-14:OAc). Samples were held for  $\geq 30$  min at room temperature or stored in a – 20 °C freezer before analysis.

TABLE 1. Location of Canadian field collections of *Ostrinia nubilalis* in 1997, 2008, 2009, and 2010, and the percentage of E and Z pheromone races or hybrids determined using gas chromatography (GC).

County/Regional Municipality	Nearest town	Geographic coordinates		Year of collection	Generation used in GC analysis	n <sup>1</sup>	Percentage		
		Latitude	Longitude				E	Z	Hybrid
<u>Ontario</u>									
Essex	Harrow	42.0021	-82.8182	2010	F12,14	28	0.0	100.0	0.0
Chatham-Kent	Ridgetown	42.2711	-81.5319	1997	F0	37	0.0	100.0	0.0
Chatham-Kent	Ridgetown	42.2711	-81.5319	2010	F14	13	0.0	100.0	0.0
Middlesex	London	42.9757	-81.1052	1997	F0	21	0.0	100.0	0.0
Huron	Brussels	43.7428	-81.2429	1997	F0	31	0.0	100.0	0.0
Niagara	Winger	42.9430	-79.4298	2008	F26	11	0.0	100.0	0.0
Niagara	Winger	42.9474	-79.3879	2010	F9	16	0.0	100.0	0.0
Leeds and Grenville	Kemptville	44.8478	-75.5509	1997	F0	21	0.0	100.0	0.0
Ottawa-Carleton	Ottawa	45.2313	-75.4337	2008	F25	14	7.1	71.4	21.4
<u>Manitoba</u>									
Grey	Elm Creek	49.7317	-98.0623	2009	F17	15	0.0	100.0	0.0
<u>Québec</u>									
Le Haut-Saint-Laurent	St. Anicet	45.1062	-74.3361	2008	F25	15	20.0	13.3	66.7

<sup>1</sup> Number of females tested with sufficient quantity of pheromone to produce GC peaks at the appropriate retention time. Peak height consisting of ≥ 95% E isomer compared to the Z isomer were classified as E-race, those with ≤ 5% E isomer were classified as Z-race, and those with intermediate percentages of E isomer (20-80%) were classified as hybrids.



Gas Chromatography

Pheromone extractions were analyzed with a Varian 3500 gas chromatograph equipped with a Varian 8200 auto-sampler (Agilent Technologies, Santa Clara, California, United States of America) using capillary techniques similar to those described by Field *et al.* (1999) and DuRant *et al.* (1995). Three  $\mu\text{l}$  of solution from the sample were injected in the gas chromatograph injector using a sandwich technique where a 0.5  $\mu\text{l}$  upper air gap was placed between the solvent plug and sample plug in a 10- $\mu\text{l}$  syringe. The air gap resides between the sample and the solvent that remains below the syringe plunger after rinsing the syringe. The air gap prevents liquid-to-liquid contact and reduces the chance of sample contamination from previous samples. A 0.8  $\mu\text{l}$  lower air gap was used to reduce sample volatilization during insertion of the needle into the hot injector.

During injection, the hot needle time was zero, the injection rate was 1.5  $\mu\text{l sec}^{-1}$ , and the needle residence time was 0.02 min. The gas chromatograph was equipped with a heated injector fitted with a 4 mm inside diameter open-top glass uniliner (Restek Corporation, Bellefonte, Pennsylvania, United States of America) containing glass wool, a fused silica capillary column (15 m x 0.25 mm) with 0.25  $\mu\text{m}$  Stabilwax® film thickness (Restek Corporation, Bellefonte, Pennsylvania, United States of America), a 5 m x 0.25 mm fused silica guard column, and a flame ionization detector. The gas chromatograph was programmed as follows: injector temp: 200 °C, splitless for 1.5 min, then set to split for the remainder of the run (split ratio 50:1 set at 60 °C); detector temp: 250 °C, attenuation set at  $32 \times 10^{-11}$ ; column oven programmed at 80 °C, held 2.0 min, heat from 80 °C to 240 °C at 10 °C min<sup>-1</sup>, held at 240 °C for 5 min to end of the run; and total run time was 23 min. Hydrogen was used as carrier gas at a flow rate of 20 cm sec<sup>-1</sup> (6.5 psi head pressure) and nitrogen was used as makeup gas. Under these conditions, the Z-7-14:OAc internal standard and the two pheromone isomers eluted at  $\approx 13.1 - 13.5$  min with each of the three peaks being separated by 0.2 to 0.4 min, which allowed for distinct separation on the chromatogram and detection of these compounds (Fig. 2).



FIGURE 1. Locations where *Ostrinia nubilalis* tested for pheromone race analysis were collected in 1997, 2008, 2009, and 2010 in Canada.

Chromatogram results for female moths for which pheromone glands were excised and analyzed by gas chromatography were categorized by pheromone race based on the percentage ratio of the two pheromone isomers. The percentages were determined by comparison of peak heights of the isomers at the appropriate retention times on the chromatogram. Samples with the peak height consisting of 95 % or more of the E isomer compared to the Z isomer were classified as E-race, those with 5 % or less of the E isomer were classified as Z-race, and those with intermediate percentages of E isomer were classified as hybrids, whereby the percentages with very few exceptions fell within the 20 % to 80 % range. Although these criteria have a broad range, analyses by C.E.M. of approximately 1000 *O. nubilalis* showed that E and Z phenotypes do not fall outside the 5 % range for the minor isomer of each phenotype (Coates *et al.* 2013). Allelic variation in a fatty-acyl reductase gene essential for pheromone biosynthesis accounts for the phenotypic variation in female pheromone production (Lassance *et al.* 2010). Mean percentages and standard deviations for the E isomer are typically about  $98.5 \pm 0.5$  % for the E race,  $67 \pm 10$  % for the hybrid, and  $3 \pm 1.0$  % for the Z race.

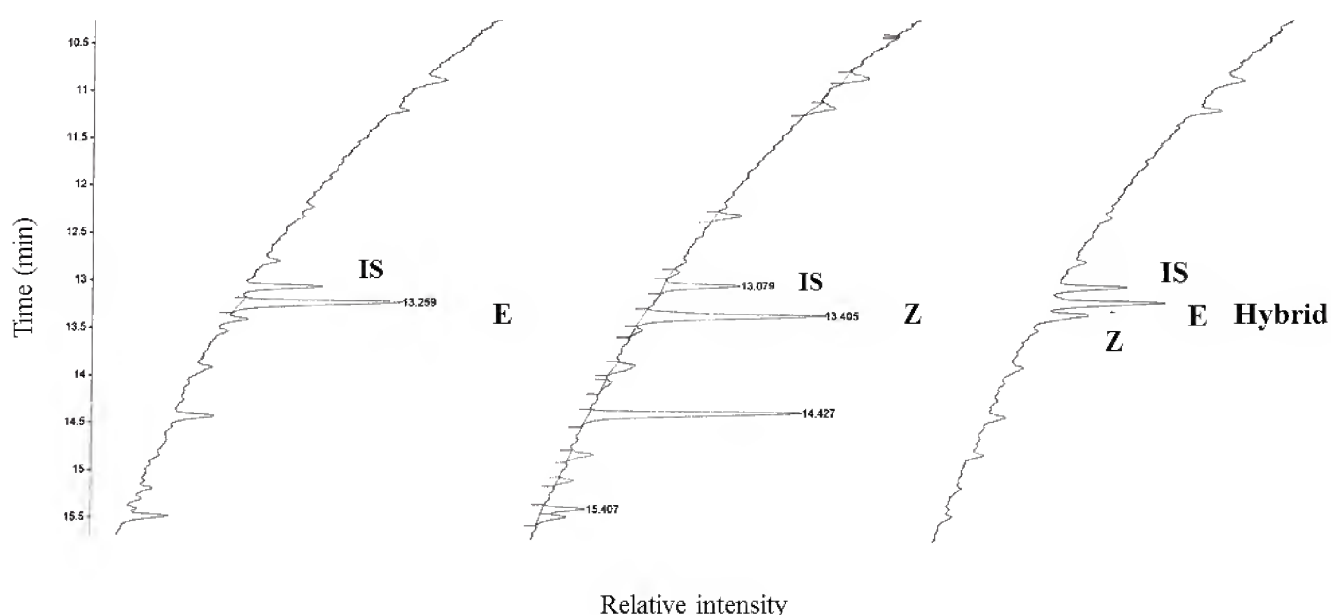


FIGURE 2. Representative chromatograms from pheromone analyses of *Ostrinia nubilalis* in Canada showing peaks for internal standard (IS), Z-11-14:Oac (Z), and E-11-14:Oac (E).

## Results and Discussion

In all years of sampling, only Z-race *O. nubilalis* were found in collections from grain corn in Essex, Chatham-Kent, Middlesex, Huron, Niagara, Leeds and Grenville Counties in Ontario, and from Grey County in Manitoba (Table 1, Fig. 1). Both E- and Z-race phenotypes were identified in samples collected in grain corn from Ottawa, Ontario and Saint-Anicet, Québec (Table 1). The Ottawa population collected in 2008 contained 21.4 % hybrid females where significant quantities of both E and Z isomers were measured by gas chromatography; however, the majority of individuals were Z-race (71.4 %) and 7.1 % were E-race (Table 1). Of the individuals tested from Québec, the majority tested were E

and Z hybrids (66.7 %), and there was a greater proportion of E-race insects (20.0 %) than Z-race (13.3 %) (Table 1).

Our results show that the Z-race of *O. nubilalis* is the dominant pheromone race found infesting grain corn in the major corn producing regions of Canada, present throughout Ontario from Essex to Ottawa Counties and in southern Manitoba. E-race *O. nubilalis* were only present in colonies derived from collections from grain corn in eastern Ontario near Ottawa and Québec, which indicates the presence of E-race within these regions. The proportion of Z or E races among founder males and females from original field collections and the resulting frequency of hybridization among offspring prior to GC analysis are unknown. Detection of E-11-14:OAc isomers in our analysis is evidence that E-race *O. nubilalis* were originally present in the area sampled. However, where no E-11-14:OAc isomers were detected there is a degree of uncertainty as to whether the E-race was lost through generations of rearing in the laboratory or because the small number of founder individuals in some collections may not have been sufficient to detect E-race individuals. A relatively new method of distinguishing pheromone races of *O. nubilalis* using SNP markers enables high throughput processing of larger sample sizes and has greater than 98 % correlation with results from GC analysis (Coates *et al.* 2013; Lassance *et al.* 2010). Therefore we are confident in our results indicating E- and Z-race phenotypes.

Klun *et al.* (1975) reported some males collected in E-race pheromone traps at Chatham and Simcoe, Ontario and St. Jean, Québec. It is possible that some E-race *O. nubilalis* were present in these areas in 1973 and 1974 when Klun *et al.* (1975) conducted their study. Our results indicate it is unlikely that the E-race is currently present in southwestern Ontario. Since we found the E-race phenotype present in the eastern range of populations we studied, it is likely that the E-race is present further east from St. Anicet, Québec and Ottawa, Ontario. Although we did not show the presence of the E-race phenotype at the Kemptville collection site, the E-race may be present there now since the samples we analyzed were from a decade earlier in the 1997 collection; however, testing of current populations must be completed for confirmation. A more in-depth study of populations of *O. nubilalis* collected from a wider host range within the regions studied would provide more conclusive information on the pheromone race composition of *O. nubilalis* in Canada. This is the first documented evidence of the E-race in Canada corresponding with the eastern corn growing region, which is a similar distribution pattern as in the U.S. These results provide useful information for pheromone trap monitoring of *O. nubilalis* in Eastern Ontario and Québec, and support observations of infestations in non-corn crops. *O. nubilalis* is also a significant pest of potato in Québec, New Brunswick, and Prince Edward Island (Noronha *et al.* 2008). Consequently, the E-race of *O. nubilalis* very well could be prominent in these areas east of where we conducted our study. Further analysis of populations collected from these regions is needed to determine this.

## Acknowledgements

The authors wish to thank John Gavloski from Manitoba Agriculture, Food and Rural Initiatives, Carman, Manitoba and Francois Meloche (retired) from Agriculture and Agri-Food Canada, Eastern Cereal and Oilseed Research Centre, Ottawa, Ontario for



sending collections of *O. nubilalis* from Manitoba, and Ottawa and Québec, respectively. We also wish to acknowledge Jennifer Bruggeman and Emily Burggraaf for their insect rearing efforts. We are grateful for the excellent anonymous review comments and suggestions.

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## THE HISTORICAL SPREAD OF THE BLACK SOLDIER FLY, *HERMETIA ILLUCENS* (L.) (DIPTERA, STRATIOMYIDAE, HERMETIINAE), AND ITS ESTABLISHMENT IN CANADA

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### Scientific Note

*J. ent. Soc. Ont.* 146: 51–54

The Black Soldier Fly, *Hermetia illucens* (Linnaeus), is a synanthropic, polysaprophagous fly native to the Neotropics, but now found in every zoogeographic region following decades of spread throughout the warmer parts of the world. In the last few decades there has been considerable interest in using larvae of *H. illucens* for organic waste control, composting, and as animal food supplements. It has been studied as an agent for manure control (Sheppard *et al.* 1994), for controlling house fly infestations in chicken production (Furman *et al.* 1959, Sheppard 1983), and as a food supplement for fish (Bondari and Sheppard 1981) and swine (Newton *et al.* 1977). More recently, there has been considerable interest in using *H. illucens* as an agent for composting (Lalander *et al.* 2015). An internet search on “Black Soldier Fly” demonstrates that this use is reaching a household level.

Although it has been suggested that *H. illucens* might have been first brought to Europe around 500 years ago (Benelli *et al.* 2014), the first verifiable Palaearctic record of the species is from southern Europe (Malta) in 1926 (Lindner 1936). The subsequent spread of this large, easily recognized species in Europe has been mainly along the Mediterranean coast of Spain, France, and Italy in the 1950s and 1960s (Leclercq 1969, 1997). In more recent years, the species has been documented spreading northwards in Central Europe: Ssymank and Doczkal (2010) recorded it from Germany and Roháček and Hora (2013) recorded it from the Czech Republic. We have seen specimens from South Africa collected as early as 1915 and from Malaysia, Hawaii, Solomon Islands, New Caledonia, Mariana Islands, Palau, and Guam in the 1940s. Probably by the 1960s, *H. illucens* had spread over most of the range it occupies today. The apparent spread of this species along coastlines and islands suggests that maritime transport may have played a role in repeated accidental introductions.

*Published December 2015*

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Although we do not know the exact original distribution of the Black Soldier Fly and it cannot be excluded that it originally occurred in the southeastern United States, its current North American range seems to reflect a northward spread from a native range in Central America and the northern parts of South America in historical times. It was present in the southern United States by the late 1800s. The earliest specimen we are aware of is from 1881 from Fernandina, Florida (United States National Museum). Riley and Howard (1889) recorded it (misspelled as *Hermetia mucens*) on the basis of a specimen collected from beehives in Alabama in 1887. We know of further records from 1897 (Louisiana), 1899 (Texas), 1911 (South Carolina), 1923 (southern California), 1926 (Virginia), 1931 (Iowa), 1938 (Ohio), 1940 (northern California), 1943 (Maryland), and 1945 (New York City). The northernmost record we are aware of is from Warner, Merrimack County, New Hampshire, 16 September 1972 (University of New Hampshire Collection). The species was not listed in works on the Diptera of Kansas (Adams, 1903) or Oregon (Cole and Lovett 1921). James (1960) shows a distribution map for California, in which the most northern specimen is at the northern end of the Central Valley. However, he also gives a North American map and mentions records from Oregon, Washington state, and North Dakota, which he discusses as “temporary local introductions”. Woodley (2001) did not include these records in his World catalog. By now, there are many records available from Oregon and Washington State, and we consider the species established in these states. One of us (NEW) did a considerable amount of collecting in southeastern Washington from about 1972 to 1980, and *H. illucens* was never seen. It now occurs there, indicating its spread in that area has been relatively recent. We know of no verified records from western Canada, the Rockies, and most of the Great Plains states.

*Hermetia illucens* is not currently recorded as occurring naturally in Canada, which is a significant issue because there is a great deal of interest in utilizing this species in Canada for waste processing, compost production, and protein production. Because Black Soldier Flies are easily reared on a wide range of decomposing materials, including animal and human waste, the large, slow-moving larvae of these flies have become popular both for manure management and pet food, and are now widely mass-reared for a variety of commercial uses in the United States, Costa Rica, Europe, and South Africa. At the moment, this species can only be imported into Canada as sterilized larvae shipped without any manure or sewage sludge (B. Gill, Canadian Food Inspection Agency, personal communication). Therefore, the question of whether or not *Hermetia illucens* occurs naturally in Canada is an important one.

There are no verifiable published Canadian records of the Black Soldier Fly, although the species is erroneously recorded from Edmonton, Alberta in the CANADENSYS database (Biodiversity Institute of Ontario, 2015). This entry is apparently an error due to contamination; there are no *Hermetia* specimens from Alberta in the Biodiversity Institute of Ontario collections (J. deWaard, Biodiversity Institute of Ontario, personal communication).

Although the Edmonton records are in error, we here record *H. illucens* from Canada for the first time on the basis of specimens collected in southern Ontario and deposited in the University of Guelph insect collection as early as 2007 (Windsor, Black Oak Savannah Park, 20 June 2007, S.M. Paiero, DEBU 002268353). This record is not surprising given the many other new Canadian records from the same area (Paiero *et al.* 2010) and given the

known occurrence of *H. illucens* in nearby Michigan. Further Black Soldier Fly specimens were documented in the course of a forensic investigation near Aurora, Ontario in August 2011 (S. VanLaerhoven, personal communication). So we can here confidently record *H. illucens* as collected repeatedly in Ontario, and it is probably established here.

Several questions remain unanswered about the history and current status of the Black Soldier Fly in Canada. The records reported here might reflect natural northward movement of this synanthropic fly, but it is more likely that the captured flies were escaped adults that emerged from imported larvae. These adults would be sterile if they hatched from legally imported sterile larvae, but is also possible that this species has been established in the province as a result of illegal importation and rearing of viable populations. Trinh *et al.* (2013), for example, indicate that their viable cultures of Black Soldier Flies originated with a supplier in Georgetown, Ontario, and it is likely that other viable cultures have been brought across the border by well-intentioned organic gardeners, fishermen, or pet owners. We suspect that the Black Soldier Fly is here to stay, but the data available to us do not allow us to say how and when it arrived.

### Acknowledgements

We thank Christopher C. Grinter, Richard S. Zack, and Robert Zuparko for checking specimens in their collections, Sherah VanLaerhoven for sharing her unpublished records, Bruce Gill for comments on an early draft of this note, Robert Walberg for encouraging us to clarify the status of Black Soldier Fly in Ontario, and Steven Paiero for depositing his first Ontario collections of Black Soldier Fly in the University of Guelph Insect Collection.

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